# APPENDIX D

**Whitpain Township**
**Naturalized Basin Maintenance Program**

The following are guidelines for maintenance of the Blue Bell Run Basin and the Valentine Estates Basin. Accompanying these guidelines are a set of plans for each basin along with information from the Pennsylvania Department of Conservation and Natural Resources for the management and control of invasive plant material. This data includes individual fact sheets for commonly found invasive herbaceous plants in Southeast Pennsylvania.

**General**

Both basins should be inspected after every major storm event and on regular biweekly basis. A record of any and all work performed at each basin should be maintained. This record will provide a basis for budgeting time and money for on-going maintenance. A draft format of a maintenance record sheet is attached for your use. At a minimum the following items should be addressed:

- Inspect the inlet and outlet structures remove any debris and note any erosion issues for future repair.

- Remove all debris from basin.

Both basins have been planted with a naturalized seed mix. The mix begins at the top of slope and, with the exception of the standing water in the Blue Bell Run Basin, covers the entire basin interior. This area should be mowed with a mulching mower on an annual basis in late February or early March as weather and ground conditions permit. The best time to mow is when the ground is frozen. Mowing height should be between 6 and 8 inches.

The remainder to the basin area (top of basin to the property line) is in either woodland or turf. The turf area should be mown on a regular basis per your normal schedule.

Inspect basin area for invasive plants on a biweekly basis in the spring and early summer (April 15 through June). Inspections for invasive plants may be reduced to a monthly basis from July 1 to first frost. Most invasive plants, if identified early, can be eradicated simply by the direct application of Roundup to the plant. Application of Roundup should be done only when the temperature is a minimum of 50 degrees and there is no forecast of precipitation for the next 8 hours. The best time to spray is in the morning hours so that the plant will have a full day of exposure to the sun. Do not spray Roundup directly into water and only spot spray the unwanted plant.

The trees and shrubs have an 18 month warranty which will expire in April 2012. Once the guarantee period is over, maintenance of the plant material will be the Township's responsibility. Except for the occasional removal of a dead branch, the trees and shrubs planted around the perimeter of the basins will not need any trimming. The plants have been selected to grow together in a hedge massing.

**Valentines Estate Basin**

- Remove any debris from the top of each of the three infiltration pits. The debris should be taken away from the basin.

**Blue Bell Run Basin**

- There is a forebay at the upper end of the basin. The purpose of the forebay is to trap sediment. It will need to be de-silted after a period of several years. The frequency will vary depending on the number and magnitude of storm events. There is a concrete pad in the bottom of the forebay. It was placed there to provide a bottom of basin for clean-out purposes. The forebay should be cleaned out when sediment is two feet above the concrete slab. The material taken from the forebay should be removed from the site.




Home
Introduction to the Tutorial
Introduction to the Problem
Invasive Plants in PA - List & Fact Sheets
Introduction to Management
Management Philosophy and Goal Setting
Inventory, Mapping and Ranking (Assesment)
Management Tools
Species Management & Control
Prevention & Early Detection
Management Planning
Monitoring & Evaluation
Restoration
Resources
Suggestions

Text only navigation and tutorial guidance questions

## Introduction to Management

**Introduction to Management**

At this stage in the tutorial, the point has well been made that invasive plants are wreaking havoc on natural lands in Pennsylvania costing tax-payers millions of dollars and diminishing our unique ecological wealth in the process (see Introduction to the Problem. The PA-DCNR Invasive Species Management Plan not only makes the issues and justification for management clear, but provides the mandate that land managers need to move forward and take action against invasive species on all of the Commonwealth's forest and park lands.

There are a number of approaches that can be taken to address invasive plant problems. The obvious considerations include money, time and available resources (tools, staff, etc.). It is most often the case that all are limited. Fortunately, there are a great many practitioners that have faced these challenges over the years and have developed systems, protocols, guidelines, plans, approaches, etc.. While others can benefit from these as they develop their own systematic systematic approach to managing and controlling invasive plants, there is . still a great deal of refining and improving that is needed. This will occur as long as land managers, are willing to document and conveypersonal experiences and information for further refinement of the process..

Given that the amount of information and variety of approaches to dealing with invasive plants is large, the challenge becomes what to include in this tutorial and when to provide a link to information . Furthermore, how to best summarize and organize the information in a way that makes it useful to you, the user, also presents its challenges.

We have organized the information presented in this tutorial within the context of the Adaptive Management Approach (AMA), a process utilized by resource managers or several decades.

Considering that each user will be coming to this tutorial with varying degrees of experience, knowledge and intent, the AMA will provide a mechanism for moving through the information from beginning to end or going directly to desired information at any desired point in the process. Other approaches, such as the Center for Invasive Plant Management's Ecological Approach to weed management exist and should be reviewed for comparison.

**Overview of the Adaptive Management Approach (AMA)**

The management of natural, biological systems is inherently difficult because prediction of ecosystem behavior is highly inexact. Predicting the outcome of invasive species management and control is difficult given that our current level of experience and knowledge is based on a relatively short history of invasive plant management. Managers are finding that taking an adaptive approach to managing invasives provides the most likely chances of success in controlling these organisms.

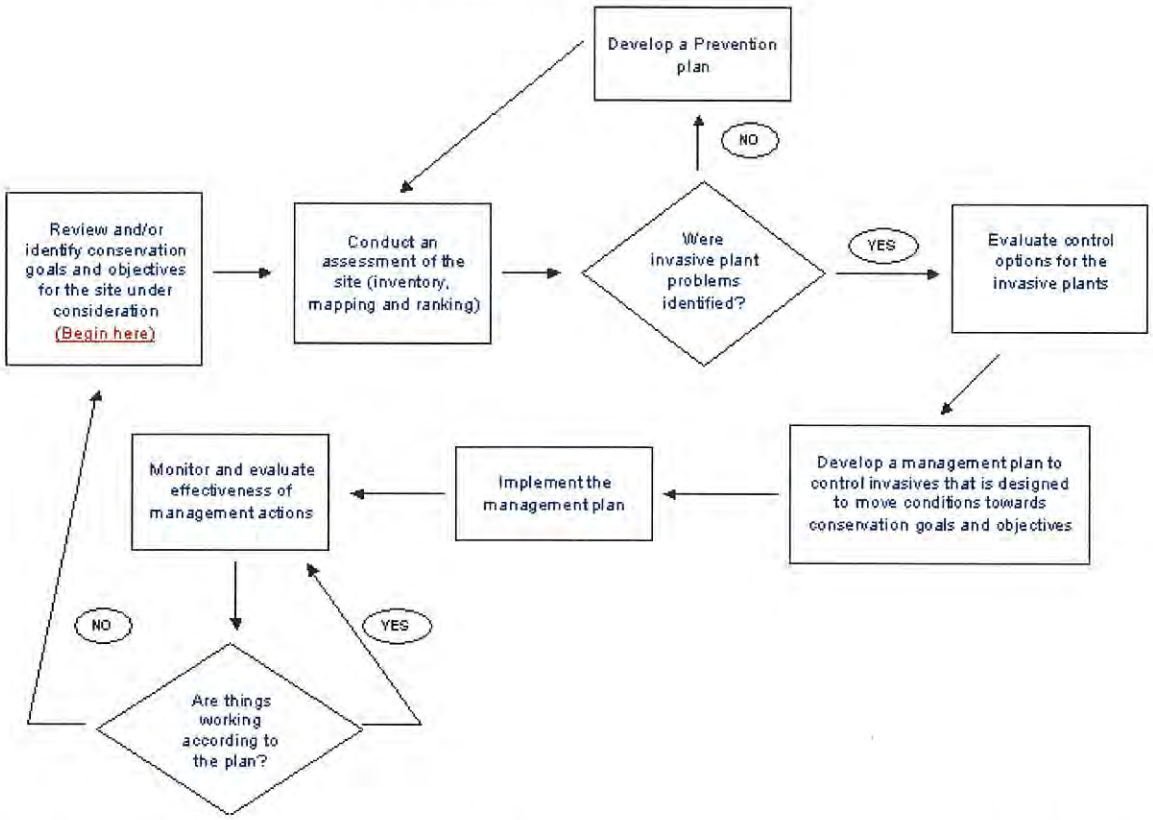
Adaptive management differs from traditional management approaches in that it allows management activities to proceed despite uncertainty regarding how best to achieve desired outcomes, and despite inevitable change and surprises. In fact, it specifically targets such uncertainty: it compels ecosystem managers to be open and explicit regarding what is not known about how best to achieve conservation and management objectives, and provides a science-based learning process characterized by using outcomes for evaluation and adjustment (Murray and Marmorek, 2003). AMA involves exploring alternative ways to meet management objectives, predicting the outcomes of each alternative based on the current state of knowledge, implementing one or more of these alternatives, monitoring to learn which alternative best meets the management objectives (and testing predictions), and then using these results to update knowledge and adjust management actions.

**AMA and Invasive Species**

The concept of adaptive management was first developed in the 1970's at the University of British Columbia and the International Institute for Applied Systems Analysis in Vienna (Holling, 1978). The Nature Conservancy and others have taken this original concept and modified it for use by land managers who are interested in addressing invasive species. To review TNC's explanation of the AMA, visit the section on Management Planning where the process is included under the TNC Management Plan template or review TNC's Weed Control Methods Handbook (Tu, M., et al., 2001) at the website, http://tncweeds.ucdavis.edu:

Before embarking on a weed management program, it is important to develop a straightforward rationale for the actions you plan to take. The following is a process diagram that takes the AMA to the next level where decision making becomes more apparent and process becomes more clear:

**Invasive Exotic Plant Management Tutorial**
**Adaptive Management Process**




(1) Establish conservation goals and objectives for the site. These may be identified in a broader management plan for the site or may need to be identified for purposes of the invasive plant management plan for the site.
(2) Determine which plant species or populations, if any, block or have potential to block attainment of the management goals and objectives by conducting an assessment. Once detected, mapping and ranking these plants becomes important.

> (a) if an assessment reveals that there are no plants or plants populations blocking attainment of goals and objectives then it is important to move into prevention mode which will require some plan of action for regularly timed assessments and a focus on early detection.

(3) Determine which methods are available to control the invasive plants.
(4) Develop and implement a management plan designed to move conditions toward management goals and objectives. Determine what control methods will work best given your site and resource constraints;
(5) Monitor and evaluate the impacts of management actions in terms of their effectiveness in moving conditions toward these goals and objectives; and

> (a) If the evaluation reveals that management goals and objectives have not been met, then begin the cycle again beginning with a determination of whether you need to modify your goals and objectives. If you reach success, then it is important to move into prevention mode which will require some plan of action for regularly timed monitoring and evaluation (or assessment).

Note that control activities are not started until the first three steps have been taken. An invasive plant control program is best viewed as part of an overall restoration program, so focus on what is desired in place of the invasive plant, rather than simply eliminating the plant. When selecting control methods, keep in mind that the ultimate purpose of the work is to preserve native species, communities, and/or functioning ecosystems.

**AMA and this Tutorial**

The management and control information in this tutorial is presented in the framework of the Adaptive Management Approach. To further explore the steps presented as part of this process, we have presented each step in the process as a Guidance Question. Information on management and control is organized in such a way as to provide you the answers to these questions. The Guidance Questions are presented on the opening page of this tutorial, as well as in the flow diagram above. This information is also organized in a Table of Contents accessible from the opening page.

## References:

Holling, C.S. (ed.) 1978. Adaptive Environmental Assessment and Management. John Wiley and Sons, New York.

Murray, C. and D. Marmorek. 2003. Adaptive Management: A Science-based Approach to Managing Ecosystems in the Face of Uncertainty. Prepared for presentation at the Fifth International Conference on Science and Management of Protected Areas: Making Ecosystem Based Management Work, Victoria, British Columbia, May 11-16, 2003. ESSA Technologies, BC, Canada.cmurray@essa.com and dmramorek@essa.com.

Tu, M., Hurd, C., & J.M. Randall, 2001. Weed Control Methods Handbook, The Nature Conservancy, Version: April 2001. http://tncweeds.ucdavis.edu > Invasives and Control Methods > Weed Control Methods Handbook.

**Additional Resources and Information on Adaptive Management Approach**

Elzinga, C.L., D.W. Salzer and J.W. Willoughby. 1998. Measuring and Monitoring Plant Populations, http://science.nature.nps.gov/im/ > Monitoring > Guidance for Designing an Integrated Monitoring Program > Invasive Species > Invasive Species Website (Invasive Species Monitoring Resources) > Monitoring Invasive Plants > Measuring and Monitoring Plant Communities (Populations). U.S. Department of Interior, Bureau of Land Management.

**Return to top of Page**




Home
Introduction to the Tutorial
Introduction to the Problem
Invasive Plants in PA - List & Fact Sheets
Text only navigation and tutorial guidance questions
Introduction to Management
Management Philosophy and Goal Setting
Inventory, Mapping and Ranking (Assesment)
Management Tools
Species Management & Control
Prevention & Early Detection
Management Planning
Monitoring & Evaluation
Restoration
Resources
Suggestions

## Management Planning

The most affective management programs have taken the time to develop a plan. Those that have been most successful are those that have been well thought out and designed to meet the goals of the project. In cases where resources (time, money and staff) are limited, management planning will ensure that those resources are used wisely. The management plan is the guiding document for any invasive plant management program and should be designed for the long-term.

Before developing a management plan under the Adaptive Management Approach, several steps need to occur first. Those include goal setting, assessment and prioritization and evaluating what methods or tools will be used to control the invasive plants that you have targeted.

To best explain the management planning process we've provided a few examples of existing management plan templates. We've also included a useful resource page from the Delaware River Invasive Plant Partnership (DRIPP) on management planning that includes useful contact information.

**Examples of Weed Management Plans**

The Nature Conservancy's Invasive Species Initiative has developed a Weed Management Plan Template that is widely used. The strength of this template is that it is self-explanatory.Measures have been taken to thoroughly explain what the user should be considering in addressing each component of the plan. The introduction is a discussion of the Adaptive Management Approach (AMA) and how the management plan fits in. Note that the AMA figure below is a more general version of that presented in the Introduction to Management. See the TNC-ISI website below (under Sources) for examples of how this template has been used.

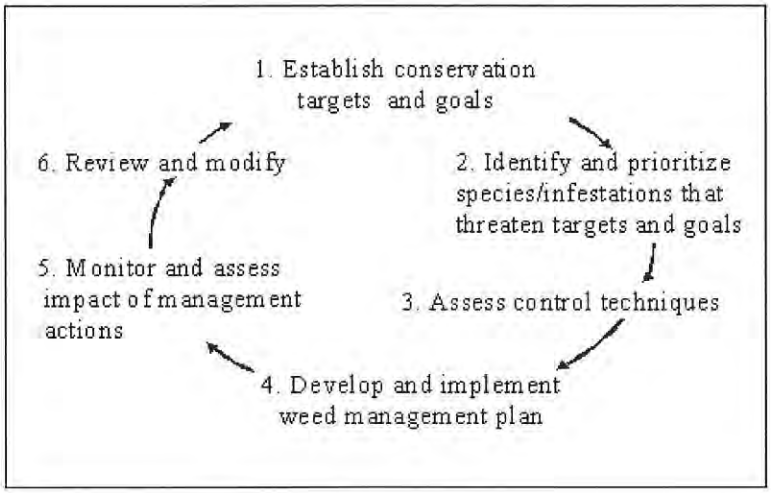
**SOURCE:** The following information on developing a management plan comes from the following source: The Nature Conservancy, Invasive Species Intiative (TNC-ISI), Weed Control Template. This template and accompanying information can be accessed at http://tncweeds.ucdavis.edu > Invasives Planning & Strategies > Templates & Examples > Weed Control Template. NOTE: This information may be updated periodically. Visit the website to check for updates.

**Using the TNC Site Weed Management Plan Template**

**A Note to Land Managers (Project Directors, Project Ecologist):**

We encourage each preserve to develop a comprehensive weed management plan. The attached Site Weed Management Plan Template is designed to make this easier. This template is also designed to help you develop an adaptive management strategy for invasive species management (see the figure below). Adaptive management of invasive species requires that you:

(1) establish management goals and conservation targets for the site;
(2) determine which, if any, species or infestations threaten or have the potential to threaten your management goals and targets - those that do are "weeds";
(3) determine which methods are available to control the weeds;
(4) develop and implement a weed management plan designed to move conditions towards the management goals and to abate threats to your targets;
(5) monitor and assess the impacts of the weed management actions in terms of the management goals and target protection;
(6) start the cycle over again by re-evaluating conclusions made in steps 1-4 and modifying where necessary.




Adaptive Management Approach

Note that you do not plan or implement weed control until steps 1-3 are completed. Step 5 is necessary to periodically re-evaluate the goals, objectives and plans made earlier so that they can be altered or modified, as called for. This is the essential "adaptive" process required by this strategy.

The template provides a standardized format with headings and boilerplate language in normal or **bold** font. Instructions and advice are in italics and can be cut as you fill in the necessary information. Advice on setting priorities for weed control is presented in Section 2A.

The computer version of the template consists of three files (all available on the internet at http://tncweeds.ucdavis.edu):
1) WMPIntro.rtf - Weed Management Plan Introduction and instructions (this file)
2) WeedTemp.rtf - Weed Management Plan template file
3) WeedTabl.xls - Tables

Also, an example of a completed weed plan from the Cosumnes River Preserve, California, is available as:

4) CRP-Plan.doc - The Cosumnes Preserve is a landscape scale project (ca 40,000 acres) and has a variety of weed problems ranging from easily treated small colonies of plants to heavily entrenched, widespread infestations of hard to control species. As a result, the Cosumnes River Preserve Weed Plan is long and relatively complicated. You should not feel compelled to develop such a large document. Rather, create the document that fits your situation and will be implemented. Do not hesitate to borrow or use text from this template, and feel free to consult with us if you have any questions about weed control methods and strategies.
5) CRP-Tabl.xls - The WeedTabl.xls spreadsheet, completed for the CRP-Plan.doc weed management plan.

**Contact us if you have any questions or comments regarding the template. Please send us copies of your completed weed management plans.**

John Randall & Barry Meyers-Rice
The Nature Conservancy
Wildland Invasive Species Program
Department of Vegetable Crops & Weed Sciences
University of California
Davis, CA 95616

Phone:
John (530) 754-8890
Barry (530) 754-8891
Fax (530) 752-4604

E-mail:
John jarandall@ucdavis.edu
Barry bazza@ucdavis.edu

http://tncweeds.ucdavis.edu

Updated July 2001 by Mandy Tu & Barry Meyers-Rice /WISP

**Site Weed Management Plan Template** - Adobe Acrobat PDF document

**SOURCE:** The following information on developing a management plan comes from the

following source: The Center for Invasive Plant Management. This information can be accessed at http://www.weedcenter.org > Weed Management Areas > Guidelines for Coordinated Management of Noxious Weeds: Development of Weed Management Areas > Section IX: Management Plan/Annual Operating Plan. Minor modifications have been made to make this information more specific to PA land managers. NOTE: This information may be updated periodically. Visit the website to check for updates.

**Management Plan and Annual Operating Plan**

Note: Weed Management Areas (WMA's) are distinguishable zones based on similar geography, weed problems, climate, or human-use patterns. These management areas cross jurisdictional boundaries between agencies, municipalities, etc. and are recognized by a natural boundary as opposed to a legal boundary. The concept is popular in the western U.S.. For purposes of the following, consider the WMA to represent your management area.

The Management Plan (MP) is the guiding document for each Weed Management Area. The Annual Operating Plan (AOP) addresses how the Management Plan is implemented on an annual basis.

A. Define/Describe the Weed Management Area
1. Name and legal description.
2. Describe boundaries.
3. Describe land use - forest, recreation, grazing, farming, mining, etc.
4. Describe topography, major aquatic features, and other natural resources.
5. Describe wildlife and flora.
6. Describe endangered species and species of special concern.
7. Identify urban areas.
8. Identify archaeological and Native American cultural sites.

B. Define Purpose of WMA Management Plan
1. Describe long-term goals, objectives, and methods for controlling noxious weeds in this WMA.
2. Identify funding and resources for weed management.
3. Establish cooperation with residents, landowners, agencies, towns, organizations, counties, and states to effectively implement programs of prevention and control within the WMA.
4. Coordinate with other WMAs in the area.

C. Define WMA Policy
1. Commitment to cooperation (See Appendix 1 for a sample cooperative agreement.)
2. Commitment to the use of Integrated Weed Management methods (See Section VII.)
3. Establish adherence to management of noxious weed in accordance with area priorities as follows:
   a. First - Prevention of potential invaders;
   b. Second - Control of new and invading species new to a particular part of the WMA; and
   c. Third - Containment and management efforts on established stands.
4. Commitment to comply with all policies for at least 5 years.

D. Define Long-Term WMA Objectives. Objectives should address the needs of the individual WMA and may not need to include all aspects of noxious weed management listed here. Also, the need for and prioritization of the following objectives will vary between WMAs. It is important to consider each of these objectives, as success is greatest when an integrated plan is developed and implemented.
1. Develop and maintain a survey and mapping system.
2. Develop and maintain funding and administration.
3. Develop awareness, education, and training programs.
4. Develop prevention and early detection programs.
5. Develop long-term management objectives for weeds of concern, according to area prioritization. (Refer to 3 above.)
6. Develop and maintain monitoring and evaluation programs.
7. Develop and maintain a reporting system.

E. Identify Weeds of Concern within the WMA.
1. List weed species and acres infested.
2. Describe methods of introduction.
3. Describe most likely areas of future infestations.

F. Develop an Integrated Weed Management (IWM) program for target weed species.
1. Describe all appropriate control methods for each weed. Use the Site Assessment Worksheet in Appendix 10 to determine the most effective IWM program.
2. Determine who will make yearly control methods recommendations. Keep recommendations current.
3. Describe safety precautions to be implemented.
4. Include corrective measures to prevent recurrence of weed infestations.

G. Define Cooperators' Roles and Responsibilities.
1. List agencies and jurisdictions involved.

2. Identify signatures required.
3. Define planning timetable.
4. Define terms and time of termination if applicable.

H. Define Collection and Management of Funds.
1. Identify sources of funding.
2. Establish a budget.
3. Determine fund management responsibilities:
a. Determine if the WMA needs its own account.
b. Determine administrative costs.

**Guidelines for Developing an Annual Operating Plan**

The Annual Operating Plan (AOP) addresses how, on an annual basis, the objectives of the over-all Management Plan are implemented. Due to manpower, funding, or other limitations, it may not be possible for the AOP to address all the objectives of the Management Plan in a given year. The Management Plan must address long term objectives and priorities. The Annual Operating Plan guides implementation of the Management Plan in yearly increments.

Budgets and circumstances may change from year to year and these changes are best addressed in making new Annual Operating Plans rather than rewriting the Management Plan annually.

An AOP may be developed for different management zones within the WMA. These zone-specific plans should be utilized only if they enhance weed management and control. For instance, a specific AOP may be necessary to manage only roadside weed problems within the WMA.

A. Roles and Responsibilities.
1. List agencies and jurisdictions involved.
2. Obtain signatures required.
3. Develop planning timetable.
4. Define terms and time of termination, if applicable.

B. Define Agreements and Compliance.
1. Voluntary agreements: compliance of all land managers within their agency guidelines.
2. Written agreements in special management areas requiring intensive management may be needed.
3. Written agreements with landowners for control of noxious weeds along roadways may be implemented.
4. Procedure for non-compliance must be followed where applicable. 5. Cooperative agreements: (See Appendix 1.)
a. Include: state agencies, municipalities, federal agencies, railroads, power company, others.
b. Should include listed noxious weed species.
6. Revegetative standards and guidelines: written plan specifying methods for accomplishing revegetation, timing, methods.
7. Cost-share programs:
a. WMA steering committee should set standards for cost-share.
b. Cost-share programs within the WMA may differ within special management areas.

C. Define Annual Funding and Resource Availability.
1. Identify sources and amount of funding.
2. Identify sources and amount of other resources:
a. Equipment availability.
b. Staff availability.
c. Cooperative mapping projects.
d. Storage availability.
e. Administration.

D. Define Specific Actions to Meet AOP Objectives
1. Implement and maintain a mapping program: (See SectionV.)
a. Define areas for survey and mapping.
b. Determine who will be responsible.
c. Determine manpower and funding required.
2. Implement prevention and early detection programs: (See Section IV.)
a. Define specific activities.
b. Determine who will be responsible.
c. Determine manpower and funding required.
3. Implement awareness, education, and training programs: (See Section III.)
a. Define specific activities.
b. Determine who will be responsible.
c. Determine manpower and funding required.
4. Implement the IWM system for the weeds of concern (See Section VII.)
a. Determine short-term IWM objectives and methods for each target weed.
b. Determine who will implement treatment program.
c. Determine manpower and funding required for control.
5. Implement and maintain monitoring and evaluation for all targeted weeds and according to Management Plan priorities and objectives (See Section VIII.)
6. Develop and maintain a reporting system for all proposed actions according to

Management Plan priorities and objectives (See Section IX.)

For examples of Management Plans and Annual Operating Plans, See Refer to Appendix 11 of The Center for Invasive Plant Management, Guidelines for Coordinated Management of Noxious Weeds: Development of Weed Management Areas which can be found at http://www.weedcenter.org > Weed Management Areas > Guidelines for Coordinated Management of Noxious Weeds: Development of Weed Management Areas > Appendix 11.

**DRIPP Management Planning Resources** - Adobe Acrobat PDF document

**Additional Resources and Information on Management Planning:**

National Invasive Species Center - http://www.invasivespeciesinfo.gov/ > Manager's Tool Kit > Control > Management Plans.

**Return to Top of Page**




Home
Introduction to the Tutorial
Introduction to the Problem
Invasive Plants in PA - List & Fact Sheets
Introduction to Management
Management Philosophy and Goal Setting
Inventory, Mapping and Ranking (Assesment)
Management Tools
Species Management & Control
Prevention & Early Detection
Management Planning
Monitoring & Evaluation
Restoration
Resources
Suggestions

Text only navigation and tutorial guidance questions

## Management Tools - Manual and Mechanical Techniques

**SOURCE:** The following information is taken from The Nature Conservancy, Invasive Species Initiative (TNC-ISI), Weed Control Methods Handbook: Tools and Techniques for Use in Natural Areas by Tu. M, C. Hurd and J.M. Randall. April 2001. This handbook can be accessed at http://tncweeds.ucdavis.edu > Invasives and Control Methods > Weed Control Methods Handbook. NOTE: This information may be updated periodically. Visit the website to check for updates.

Manual and mechanical techniques such as pulling, cutting, and otherwise damaging plants, may be used to control some invasive plants, particularly if the population is relatively small. These techniques can be extremely specific, minimizing damage to desirable plants and animals, but they are generally labor and time intensive. Treatments must typically be administered several times to prevent the weed from re-establishing, and in the process, laborers and machines may severely trample vegetation and disturb soil, providing prime conditions for re-invasion by the same or other invasive species.

Manual and mechanical techniques are generally favored against small infestations and/or where a large pool of volunteer labor is available. They are often used in combination with other techniques, for example, when shrubs are pulled and cut, and re-sprouts and seedlings are treated with herbicides or fire several weeks or months later.

When using manual and mechanical methods, it is especially important to thoroughly clean and inspect all equipment and clothing before moving it off-site. This will lessen the probability of spreading the weed (s) to the next worksite.

In addition to the tools described here, the Wildland Invasive Species Team web page reviews other innovative tools. See tncweeds.ucdavis.edu/tools.html

### A. WEED PULLING

Pulling or uprooting plants can be effective against some shrubs, tree saplings, and herbaceous and floating weeds. Annuals and tap-rooted plants are particularly susceptible to control by hand-pulling. Weed wrenches and other tools are surprisingly powerful and can enable you to control large saplings and shrubs that are too big to be pulled by hand. It is not as effective against many perennial weeds with deep underground stems and roots that are often left behind to re-sprout.

**How To**: Minimize soil disturbance by pulling out weeds slowly and carefully, and replace soil to disturbed areas where possible. Trampled and disturbed areas can provide optimal germination sites for many weeds. Minimize trampling by limiting the number of people in the site and the amount of time spent there. Whenever a manual technique is used, it is wise to wear gloves, a long-sleeved shirt, and long pants. Some plants can cause moderate to severe skin irritation, especially when their stems and leaves are crushed and broken. Even the flimsiest weeds can leave hands raw and bleeding after several hours of pulling.

The advantages of pulling include its small ecological impact, minimal damage to neighboring plants, and low (or no) cost for equipment or supplies. Pulling is extremely labor intensive, however, and is effective only for relatively small areas, even when abundant volunteer labor is available.

#### 1. Hand Pulling

Hand pulling is easy to plan and implement, and is often the best way to control small infestations, such as when a weed is first detected in an area. Hand pulling may be a good alternative in sites where herbicides or other methods cannot be used. The key to effective hand pulling is to remove as much of the root as possible while minimizing soil disturbance. For many species, any root fragments left behind have the potential to re-sprout, and pulling is not effective on plants with deep and/or easily broken roots.

Hand pulling has been effective against a variety of invaders in natural areas scattered across the U.S. For example, hand pulling by volunteers has successfully controlled Centaurea diffusa (diffuse knapweed) in the Tom McCall Preserve in northeast Oregon. Yellow bush lupine (Lupinus arboreus) was also controlled in coastal dunes in California by pulling small shrubs by hand. Larger shrubs were cut down with an ax, and re-sprouting was uncommon (Pickart and Sawyer 1998). Hand pulling has also been fairly successful in the control of small infestations of Centaurea spp. (thistles), Melilotus officinalis (white and yellow clover), and Lythrum salicaria (purple loosestrife) at TNC preserves scattered across the country.

**2. Pulling Using Tools**

Most weed-pulling tools are designed to grip the weed stem and provide the leverage necessary to pull its roots out. Tools vary in their size, weight, and the size of the weed they can extract. The Root Talon is inexpensive and lightweight, but may not be as durable or effective as the all-steel Weed Wrench, which is available in a variety of sizes. Both tools can be cumbersome and difficult to carry to remote sites. Both work best on firm ground as opposed to soft, sandy, or muddy substrates.

**Root Talon**
The Root Talon is an inexpensive and lightweight tool shaped something like a pick-ax with a plastic handle and metal head. It has a specialized claw and gripping device that allow the user to grab the plant stem and provide leverage to pull-up and remove the plants. It is best used for pulling shallow rooted plants such as sapling trees and herbs with sturdy stems. Plants that have been pulled using the Root Talon include young tree-of-heaven (Ailanthus), Scarlet wisteria (Sesbania punicea), and buckthorn (Rhamnus spp.). The Root Talon is not effective against deep-rooted plants, because it does not provide enough leverage. In addition, it is difficult to use the Root Talon to pull spiny plants because the plant stems (and spines) must be put into the gripping flange by hand. Advantages of the Root Talon are that it is lighter and less expensive than the Weed Wrench (see below), and provides easier and more effective control than hand pulling.

At the time of printing, the Root Talon retailed for $47 plus $5.25 shipping through Lampe Design, LLC, 262 South Griggs Street, St. Paul, MN 55105, (612) 699-4963, jklampe@worldnet.att.net or on the web at www.buckthorn.com.

**Weed Wrench**
The Weed Wrench provides more leverage than the Root Talon. Its all-steel frame is capable of withstanding more strain than the plastic handle of the Root Talon. It comes in four sizes, from the "mini", which weighs 2.4 kg (5.25 lbs) and is capable of pulling weeds with stems up to 2.5 cm (1.0 in) in diameter, to the "heavy", which weighs 10.5 kg (24 lbs) and can handle weeds up to a diameter of 6.25 cm (2.5 in). Larger Weed Wrenches provide more leverage and pulling power. It is best to choose the smallest size needed, however, because larger Weed Wrenches are heavy and can be difficult to carry and use in remote sites.

Manufacturers of the Weed Wrench claim it is capable of handling any plant that can fit within the "jaws" of the wrench, as long as the plant stem is stronger than the anchoring strength of the roots. The Weed Wrench can be used on herbaceous plants that have a stem or bundle of stems strong enough to withstand the crush of the jaws. It has been used successfully to pull acacia (Acacia melanoxylon), buckthorn (Rhamnus cathartica), Russian olive (Elaeagnus angustifolia), multiflora rose (Rosa multiflora), willow (Salix spp.), tamarisk (Tamarix spp.), bush honeysuckles (Lonicera spp.), Scotch broom (Cytisus scoparius), French broom (Genista monspessulanus), and Brazilian pepper (Schinus terebinthifolius) at preserves across the mainland U.S. In Hawaii, the Weed Wrench has been used to pull Strawberry guava (Psidium cattleianum) and small saplings of Karaka nut (Corynocarpus laevigatus) from the Kamakou preserve on Molokai (Hawaii).

For more information, contact The Weed Wrench Company, at 2852 Willamette Street #403, Eugene, OR 97405, 1-877-484-4177, connect@weedwrench.com. You can also view their website at http://www.weedwrench.com.

**B. MOWING, BRUSH-CUTTING, WEED EATING**
Mowing and cutting can reduce seed production and restrict weed growth, especially in annuals cut before they flower and set seed (Hanson 1996). Some species however, re-sprout vigorously when cut, replacing one or a few stems with many that can quickly flower and set seed. For example, yellow starthistle (Centaurea solstitialis) can be controlled by mowing at the onset of flowering (when approximately 2 to 5% of the seed heads are flowering), but if mowed earlier, native species are negatively impacted and yellow starthistle is able to re-sprout (Benefield et al. 1999). Be sure to consider the biology of the weed before cutting.

**How To:** Mowing and cutting are often used as primary treatments to remove aboveground biomass, in combination with prescribed burning or herbicide treatments. It is important to collect the cut fragments of species capable of re-sprouting from stem or root segments to prevent them from washing or blowing into uninfested areas.

**C. STABBING**
Some plants can be killed by severing or injuring (stabbing) the carbohydrate storage structure at the base of the plant. Depending on the species, this structure may be a root corm, storage rhizome (tuber), or taproot. These organs are generally located at the base of the stem and under the soil. Cutting off access to these storage structures can help "starve" or greatly weaken some species.

**How To:** To sever a taproot, place a flat-nosed spade, pruning saw, or knife at the base of the plant and push it as far below ground as possible. To prevent re-sprouting, the taproot should be severed below the caudex or root crown (where the stem becomes the root).

The stabbing technique has been used to control baby's breath (Gypsophila paniculata) in Michigan (J. McGowan-Stinski, pers. comm.). The stabbing of root corms has also been an effective control technique for large (two yr old) plants of burdock (Arctium lappa) and wild parsnip (Pastinaca sativa) in Illinois and Wyoming (W. Kleiman, pers. comm.).

**D. GIRDLING**
Girdling is often used to control trees or shrubs that have a single trunk. It involves cutting away a strip of bark several centimeters wide all the way around the trunk. The removed strip must be cut deep enough into the trunk to remove the vascular cambium, or inner bark, the thin layer of living tissue that moves sugars and other carbohydrates between areas of production (leaves), storage (roots), and growing points. This inner cambium layer also produces all new wood and bark.

**How To:** To girdle a tree, cut parallel lines approximately three inches or more apart around the circumference of the tree. The cuts can be made using a knife, ax, or saw, and should be slightly deeper than the cambium. Strike the trunk sharply between the cuts using the back of an ax or other blunt object. The bark should come off in large pieces and prevent the tree from any further growth. It is important not to cut too deeply into the trunk because this could cause the tree to snap and fall in high winds. To determine the depth of the cambium, make two short test cuts and strike the bark between the cuts. After several strikes the bark should come off intact, exposing the cambium and wood (xylem) below.

Girdling is effective against pines, some oaks, and some maples. It typically requires less labor than cutting and removal, is inexpensive, and kills

only the targeted plant. It also leaves no residue except the standing trunks. In addition, a dead standing tree (snag) can provide valuable wildlife habitat, and if left to decay, allows the nutrients of the tree to be returned to the system, rather than being removed and deposited elsewhere. A few species, notably black locust (Robinia pseudoacacia) and tree of heaven (Ailanthus altissima) should not be girdled because they respond by producing many fast growing root and stem sprouts. Therefore, before girdling, find out if the target species responds by re-sprouting. If so, use another control technique, such as hack and squirt herbicide applications or if you do girdle return at 1 to 4 month intervals to cut, burn, or herbicide all re-sprouts for at least 2 years.

Girdling has been used successfully on preserves in New York state to control quaking aspen (Populus tremuloides) and bigtooth aspen (Populus grandidentata). Girdling can also be used in combination with herbicides. Black locust (Robinia pseudoacacia) and quaking aspen (P. tremuloides) in New York and Wisconsin, respectively, were controlled successfully using girdling with herbicide. This method, however, was not successful, in controlling tropical ash (Fraxinus uhdei) on the Kamakou preserve on Molokai, Hawaii.

**E. MULCHING**
Mulching can be used on relatively small areas, but will often stunt or stop growth of desirable native species. Mulching cannot control some perennial weeds because their extensive food reserves allow them to continue to grow up through the mulch.

**How To:** Cover the ground and/or seedlings with mulch (hay, grass clippings, wood chips, etc.) or other type of ground cover (newspaper clippings). This prevents weed seeds and seedlings from receiving sunlight necessary to survive and grow.

Hay mulch was used in Idaho with some success to control the spread of Canada thistle (Cirsium arvense). This hay mulch was applied several feet deep to established plants, and even though these plants were not completely eliminated, flowering rates were much suppressed by the end of the growing season.

**F. TILLING**
Tilling, or the turning-over of soil, is often used for weed control in agricultural crops. Its use in wildland management is largely limited, however, to restoration sites where soils are already badly disturbed. Tilling is effective against annuals and shallow-rooted perennials, but small fragments of some species, particularly those perennials with rhizomes, can often resprout following tillage. Tilling should be completed before seeds develop and are shed onto the soil. The best control is achieved when the soil remains dry, so that remaining plant fragments dry out. Moist soils help the fragments survive and re-grow.

**How To**: "Primary" tillage equipment is initially used to turn over soil and cuts roots at depths of six inches to two feet to prepare the soil for planting. "Secondary" tillage equipment, or equipment designed to work only the top six inches of soil, is used mainly to control weeds.

Many types of secondary tillage equipment are available. Equipment ranges from small hand-pushed models, to tractor mounted power-driven tillers. The appropriate model depends on the size and type of the habitat.

**G. SOIL SOLARIZATION**
Soil solarization is the technique of placing a cover (usually black or clear plastic) over the soil surface to trap solar radiation and cause an increase in soil temperatures to levels that kill plants, seeds, plant pathogens, and insects. In addition, when black plastic or other opaque materials are used, sunlight is blocked which can kill existing plants (Katan et al. 1987). Soil solarization however, can cause significant biological, physical, and chemical changes in the soil that can last up to two years, and deter the growth of desirable native species.

Soil solarization is used in horticulture and for a few high value agriculture crops like strawberries. This method has not been used extensively for weed control in natural settings. The effectiveness of soil solarization depends, in part, on how susceptible weed seeds are to temperature increases. It is most effective against winter annual weeds that germinate under cool conditions (Elmore 1990). Summer annuals and other species adapted to higher temperatures, which germinate during warmer parts of the year, are less susceptible.

Soil solarization is most effective during the summer months, and may be less effective in cooler climates (DeVay 1990). The higher the temperature, the more quickly a kill is achieved. Solarization is effective only if done in wet soil. Where soils are typically dry, they must first be irrigated until soil from the surface to 50 to 60 cm deep is at field capacity (Grinstein & Hetzroni 1991).

**How To**: Polyethylene plastic film is the most useful for soil solarization (DeVay 1990). Less expensive thin films (1-1.5 mil) are more effective than thick films (2, 4, and 6 mil). Clear and black films both trap infrared radiation that is re-radiated from the soil surface, therefore keeping the soil hot. Transparent film allows more radiation to reach the soil than black films, as it lets visible light in, causing even greater temperature increases. Because black films exclude visible light however, they stop photosynthesis, which can be enough to kill some young annuals and perennials given sufficient time (Elmore 1990). Double layers of film have been found to increase soil temperatures by three to ten degrees over single layers (DeVay 1990).

Soil solarization is beneficial in that it releases nutrients that are tied up in the organic component of the soil, and that it can kill unwanted plants without the use of chemicals (Stapleton 1990). However, solarization leaves an open substrate that can be readily invaded by new organisms, both native and non-native once the plastic is removed (Stapleton 1990). The influx of nutrients that results from solarization can be advantageous to restoration efforts, but can promote aggressive, ruderal plants that typically thrive in nutrient-rich soils.

**H. FLOODING**
In situations where the water level of a wetland or riverine system can be manipulated, flooding can be used to control some plant species. Some species, however, have vegetative buds or underground storage organs that can survive several months or more under flooded conditions.

In Vermont, flooding was used successfully to kill seeds and seedlings of common buckthorn (Rhamnus cathartica). Flooding was also used in combination with herbicide to successfully control the spread of autumn olive (Elaeagnus umbellata) and reed canarygrass (Phalaris arundinacea) in Ohio. At Wertheim NWR on Long Island, NY, Phragmites australis was controlled by burning and then flooding with several feet of water in impounded areas.

## References

Benefield, C.B., DiTomaso, J.M., Kyser, G.B., Orloff, S.B., Churches, K.R., Marcum, D.B., and G.A. Nader. 1999. Success of mowing to control yellow starthistle depends on timing and plants branching form. California Agriculture 53(2): 17-21.
DeVay, J.E. 1990. Historical review and principles of soil solarization. In: DeVay, J.E., Stapleton, J.J., and C.L. Elmore (eds.), Soil Solarization. United Nations, Rome.
Elmore, C.L. 1990. Use of solarization for weed control. In: DeVay, J.E., Stapleton, J.J., and C.L. Elmore (eds.), Soil Solarization. United Nations, Rome.
Grinstein, A. and A. Hetzroni. 1991. The technology of soil solarization. In J. Katan and J.E. DeVay (eds.) Soil Solarization. CRC Publications, Boca Raton: 159-170.
Hanson, E. 1996. Tools and techniques. Chapter 3 in Invasive plants. J. M. Randall and M. Marinelli, eds. Handbook #149. Brooklyn Botanical Garden, Inc., Brooklyn, New York. 111 pgs.
Katan, J., Grinstein, A., Greenberger, A., Yarden, O. and J.E. DeVay. 1987. First decade (1976-1986) of soil solarization (solar heating)-A chronological bibliography. Phytoparasitica 15:229-255.
Pickart, A.J. and J.O. Sawyer. 1998. Ecology and restoration of Northern California coastal dunes. California Native Plant Society. Sacramento, CA. 152 pgs.
Stapleton, J.J. 1990. Thermal inactivation of crop pests and pathogens and other soil changes caused by solarization. In: DeVay, J.E., Stapleton, J.J., and C.L. Elmore (eds.), Soil Solarization. United Nations, Rome.

**Date Authored:** April 2001

**Return to top of Page**

# DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers




- Home
- Introduction to the Tutorial
- Introduction to the Problem ➤
- Invasive Plants in PA - List & Fact Sheets
- Introduction to Management ➤
- Management Philosophy and Goal Setting
- Inventory, Mapping and Ranking (Assesment) ➤
- Management Tools ➤
- Species Management & Control
- Prevention & Early Detection ➤
- Management Planning ➤
- Monitoring & Evaluation ➤
- Restoration ➤
- Resources ➤
- Suggestions

Text only navigation and tutorial guidance questions


## Management Tools - Chemical

**SOURCE:** The following information is taken from The Nature Conservancy, Invasive Species Initiative (TNC-ISI), Weed Control Methods Handbook: Tools and Techniques for Use in Natural Areas by Tu. M, C. Hurd and J.M. Randall. April 2001. This handbook can be accessed at http://tncweeds.ucdavis.edu > Invasives and Control Methods > Weed Control Methods Handbook. NOTE: This information may be updated periodically. Visit the website to check for updates.

**Guidelines for Herbicide Use**

**PURPOSE**

These Guidelines are designed to ensure that you carefully consider the overall impacts of herbicide use on your conservation targets, other native species, and the ecological system. Base all decisions whether to control weeds, and whether to use herbicides instead of other methods, on the conservation targets and management goals for the site. In addition, the health and safety of applicators and others in the vicinity must be considered BEFORE pesticides are applied. Simply put, one should be confident that the proposed herbicide will do more conservation good than harm and not endanger the health of the applicators or others in the area.

**TO SPRAY OR NOT TO SPRAY?**

Determining the right course of action in weed management can be difficult. For many land managers, whether to apply herbicides is an ethical decision that is not taken lightly. Herbicides are often used as a last resort, when other attempts have failed, and action is imperative.

The following checklist summarizes the steps that need to be taken to ensure that proper consideration has been given to current weed problems, and that the use of herbicides is warranted for each individual case.

1. Determine whether invasive plants threaten conservation targets or management goals on the site. Use herbicides (versus other control methods) only if confidant they can be used safely and will do more conservation good than harm. If you decide to use herbicides, be sure to record your reasons for doing so. TNC's Site Conservation Program (http://www.consci.org/scp) can help you identify targets and threats, and make a Site Conservation Plan. TNC's Site Weed Management Plan Template (http://tncweeds.ucdavis.edu/products.html) can help you set control priorities and develop a plan to implement them.
2. Develop safety protocols for STORING, MIXING, TRANSPORTATING, HANDLING SPILLS, and DISPOSING OF UNUSED HERBICIDES & CONTAINERS BEFORE obtaining herbicides.
3. Follow all federal, state and local regulations regarding herbicide use. You MUST read and follow product labels. It is a violation of federal law to use an herbicide in a manner inconsistent with its label.
4. Contact your State Department of Agriculture or County Agriculture Commissioner for information about state and local regulations regarding applicator permits and posting requirements. (See the list of state regulatory agencies in the Appendix.)
5. Check with the legal staff for your program (State or Regional Office) BEFORE obtaining herbicides if you have any questions about regulations or liability issues.
6. Herbicides may be applied only by TNC employees or contractors who have all certificates and licenses required by the state and/or county. Volunteers may NOT apply herbicides unless they are properly licensed AND have signed a consent & release form.
7. Applicators MUST wear all protective gear required on the label of the herbicide they are using. Provide all safety and protective gear requested by the employee(s) applying the herbicide. The health and safety of the applicator are of foremost concern.

These Guidelines and TNC's Standard Operating Procedures were designed to make TNC use of herbicides meet or exceed the Worker Protection Standard for Agricultural Pesticides enacted by the U.S. EPA January 1 1995. Although the Worker Protection Standard does not cover pesticide use in natural areas, except on sites leased for agricultural production, TNC's operations should at the very least measure up to this Standard. It is NOT the purpose of TNC's Standard Operating Procedures nor of these Guidelines to require stewards to produce lengthy herbicide use plans.

**SITE CONDITIONS**

Site conditions to be considered include accessibility, proximity to open water, depth to groundwater, the presence of rare species and other conservation targets, and the site's sensitivity to trampling that could occur when the herbicide is being applied.

To prevent contamination of water bodies, management plans should carefully consider the hydrology of the system that is being treated. Hypothesize potential runoff scenarios and take appropriate measures (such as buffer zones) to prevent them. Underground aquifers and streams should be considered as well.

The herbicides covered in this Manual are regarded as posing relatively low risk for use in natural areas because they are not likely to contaminate groundwater, have limited persistence in the environment, and are of low toxicity to animals. Critical reviews of several common herbicides are available at a small charge from the Northwest Coalition for Alternatives to Pesticides (NCAP, P.O. Box 1393, Eugene, OR 97440, (503) 344-5044, http://www.pesticide.org). Information is also available from the National Coalition Against the Misuse of Pesticides (NCAMP, 701 E Street SE #200, Washington DC 20003, (202) 543-5450, http://www.beyondpesticides.org/).

In addition to federal pesticide registration, some states also have their own registration procedures and requirements and almost all states have their own pesticide applicator licensing, certification, or registration. To find out if a particular herbicide is registered for use on wildlands in your state, call the state pesticide regulatory agency (see the Appendix for a list of state regulatory agencies).

**ENDOCRINE DISRUPTING COMPOUNDS**

The presence of synthetic chemicals in the environment, especially those designed to control unwanted species (insecticides and herbicides), and the acute and long-term effects of those chemicals on wildlife and humans have been of concern since the publication of Rachel Carson's book "Silent Spring" in 1962. New evidence indicates that the functioning of animals (including humans) endocrine systems can be severely altered by low-level cumulative exposure to some synthetic chemicals. Many different classes of industrial chemicals released into the environment exhibit potential endocrine-disrupting activities, such as mimicking or blocking the action of natural animal hormones. Exposure to these compounds during critical periods of development (in utero, or early postnatal) can result in irreversible damage to wildlife and to humans. In general, the compounds found in insecticides are usually more toxic than those in most herbicides, as most herbicides block or alter biochemical processes found exclusively in plants.

Numerous studies have reported that agricultural and industrial waste chemicals adversely effect wildlife populations. Endocrine-altering compounds, however, can also be found naturally (such as the phytoestrogen genistein, that is found in soy protein). Some studies suggest that the effects of synthetic chemicals are negligible relative to those of naturally occurring plant estrogens. Many synthetic compounds are known to bioaccumulate, which may greatly magnify their effects. It has also been suggested that combinations of synthetic compounds act synergistically with effects far greater than those of any one compound.

Some studies suggest that synthetic endocrine-disrupting chemicals alter growth, development, and reproduction rates, and can cause abnormal behavior in various wildlife species. Further, there is increasing concern regarding potential effects of synthetic endocrine disruptors on human reproduction and development, including, but not limited to, increased breast and ovarian cancers, infertility, increased testicular cancer, decreased semen quality, and increased spontaneous abortion rates.

A review by CAST (Council for Agricultural Science and Technology) published in 2000, concluded that current scientific evidence does not clearly link endocrine-disrupting chemicals with decreased male reproductive capacity or increased rates of breast cancer in women. However, this review did not completely dismiss the potential role that these chemicals may have as causative agents for adverse human health effects. Herbicides are only a small subset of all synthetic chemicals produced, and thus far, only 2,4-D has been implicated for possible endocrine-disrupting impacts. Some reproductive and developmental problems in wildlife populations have been attributed to endocrine-disrupting chemicals, but evidence of other effects are far from conclusive.

**For more information:**

Colborn, T., Dumanoski, D. and J.P. Myers. 1996. Our Stolen Future: Are We Threatening Our Fertility, Intelligence and Survival. A Scientific Detective Story. Penguin Books, New York.

Cornell University Program on Breast Cancer and Environmental Risk Factors in New York State. 2000. Endocrine Disruption and Breast Cancer Risk. http://envirocancer.cornell.edu/Bibliography/General/bib.endocrineDisruption.cfm

Lyons, G. 1999. Endocrine disrupting pesticides. Pesticides News 46: 16-19. Pesticide Action Network UK.

Safe, S.H., Foster, W.G., Lamb, J.C., Newbold, R.R. and G. Van Der Kraak. 2000. Estrogenicity and endocrine disruption. Council for Agricultural Science and Technology (CAST), Issue Paper no. 16.

**HERBICIDE PROPERTIES**

Consider the following herbicide properties when deciding which compound to use:

1. Effectiveness against the target species.
2. Mechanisms of dissipation (persistence, degradation, and likelihood of movement via air or water to non-target organisms).
3. Behavior in the environment (in soils, water, and vegetation).
4. Toxicity to birds and mammals, aquatic species, and to other non-target organisms (including algae, fungi, and soil organisms).
5. Application considerations
6. Safety
7. Human toxicology

In general for work in natural areas, it is best to select compounds that are effective against the weed, not likely to drift, leach to groundwater or wash into streams, nontoxic to people and other organisms, not persistent in the environment, and is easy to apply. In some circumstances, a single application of a more toxic or persistent chemical that kills the weed, however, may be preferable to a less persistent, less toxic compound that must be applied repeatedly. Strive to do the job with the smallest total negative impact to the environment.

**PROTECTIVE GEAR FOR APPLICATORS**

The health and safety of the applicator are of foremost concern. Applicators MUST wear all protective gear required on the label of the herbicide they are using. Any additional safety and protective gear requested by TNC applicators must be provided. See the following textbox (page 5.6) for additional information regarding personal protection needs.

Even if not required, all TNC or volunteer applicators should wear the following when mixing or applying herbicides:

1. Rubber boots,
2. Protective aprons or suits (e.g., disposable tyvek suits) or sturdy overalls that are not used for other activities,
3. Rubber gloves (tyvek and nitrile gloves are recommended - one study indicated that neoprene can be penetrated by herbicides under field conditions),
4. Safety glasses or goggles.

Some applicators may even wish to wear respirators where not required. A dust mask may be worn when a respirator is not required, but pesticide safety officers point out that dust masks usually fit loosely and do not stop volatile compounds. Furthermore, they can indirectly increase chances of exposure if they cause heating, sweating, and irritation, which induce the wearer to repeatedly wipe or scratch their face.

Some companies that supply protective gear include:

A.M. Leonard, Inc.
241 Fox Drive
Piqua, Ohio 45356-0816
Phone: 1-800-543-8955
Web Address: www.amleonard.com

Ben Meadows Company
190 Etowah Industrial Court
Canton, GA 30114
Phone: 1-800-241-6401
Web Address: www.benmeadows.com

Forestry Suppliers, Inc.
P.O. Box 8397
Jackson, MS 39284-8397
Phone: 1-800-647-5368
Web Address: www.forestry-suppliers.com

Gempler's Inc.
P.O. Box 270
Belleville, WI 5350
Phone: 1-800-382-8473
Web Address: www.gemplers.com

Lab Safety Supply Inc.
P.O. Box 1368
Janesville, WI 53547-1368
Phone: 1-800-356-0783
Web Address: www.labsafety.com

Safety Solutions, Inc.
6161 Shamrock Ct.
P.O. Box 8100
Dublin, Ohio 43016-2110
Phone: 1-800-232-7463
Web Address: www.safetysolutions.com

## Personal Protection in Herbicide Handling

Adapted from Ohio State University's Extension Publication #825 "Applying Pesticides Correctly" by Jennifer Hillmer, The Nature Conservancy-Ohio

**PERSONAL PROTECTIVE EQUIPMENT**
Herbicide labels indicate the minimum protective equipment required. This may vary by application technique. Cotton, leather, canvas, and other absorbent materials are not chemical resistant, even to dry formulations.

- Always wear at least a long-sleeved shirt, long pants, sturdy shoes or boots, and socks. The more layers of fabric and air between you and the pesticide, the better the protection.
- A thick layer of spray starch on clothing will add some protection from pesticides.
- Hands and forearms usually receive the most pesticide exposure. Wear chemical-resistant gloves, and tuck shirt sleeves into gloves (gloves should reach up the forearm, with cuffs to catch runs and drips).

- Canvas, cloth, and leather shoes or boots are almost impossible to clean adequately. Wear chemical-resistant rubber boots that come up at least halfway to the knee if the lower legs and feet will be exposed to herbicides or residues.

**AVOIDING CONTAMINATION**

- Wear chemical-resistant gloves (rubber or plastic such as butyl, nitrile, or polyvinyl chloride are common types).
- Make sure gloves are clean, in good condition, and worn properly. Replace gloves often. Wash and dry hands before putting on gloves. Wash gloves before removing them.
- Wash hands thoroughly before eating, drinking, using tobacco products, or going to the bathroom.
- Cuff gloves if pesticide is expected to run down towards the sleeves. Tuck sleeves into gloves.

**EYE AND RESPIRATORY PROTECTION**

- PPE labeling might require goggles, face shields, or safety glasses with shields. Some formulas or handling activities pose more risks to eyes than others. Dusts, concentrates, and fine sprays have the highest risk of causing pesticide exposure.
- There are many types of dust-mist masks and respirators, all of which must fit and be used properly to be effective.
- Respiratory protection is most important in enclosed spaces or when the applicator will be exposed to pesticides for a long time.
- Pesticides that can volatilize require the use of respirators. Check label requirements.

**PERSONAL CLEAN-UP AFTER HERBICIDE USE**

- Wash gloves and footwear (if possible) with detergent and water before removing them.
- Change clothing and put clothes used during application in a plastic box or bag, and keep it away from children or pets Use a mild liquid detergent and warm water to wash your hands, forearms, face, and any other body parts that may have been exposed to pesticides. Take a warm shower and wash your hair and body at the end of the work day.

**LAUNDRY**

- Do not wash work clothing and personal protective equipment in the same wash water with the family laundry. Handle with care and wash your hands after loading the machine.
- If you have chemical-resistant items, follow the manufacturer's washing instructions. Wash boots and gloves with hot water and liquid detergent. Wash twice, once outside and once inside. Air-dry boots and gloves.
- Rinse clothes in a machine or by hand.
- Wash in plenty of water for dilution and agitation.
- If using a washing machine, using heavy-duty liquid detergent in hot water for the wash cycles.
- After washing the clothes, run the washer through one complete cycle with detergent and hot water, but no clothing, to clean the machine.
- Hang items to dry if possible in plenty of fresh air. Do not hang in living areas.
- Using a clothes dryer is acceptable, but over time the machine may become contaminated with pesticide residues.

**EMERGENCY PRECAUTIONS AND EQUIPMENT**

Applicators must have easy access to emergency decontamination and first aid kits whenever they are applying herbicides, even if they are out in the field. All applicators should have access to an eyewash kit and at least 2 gallons of clean water.

Decontamination kits are available from many suppliers or can be assembled independently. Rubber buckets or tubs with tight sealing lids are convenient for homemade kits and should include:

1. Two (or more) 1 gallon containers filled with potable water,
2. Eyewash kits or eyewash bottles with buffered isotonic eyewash,
3. Hand or body soap (bring enough for all workers to thoroughly wash their hands when in the field),
4. Paper or other disposable towels,
5. A full tyvek coverall with foot covers,
6. A map and directions to the nearest medical facilities. Such maps should be posted in prominent locations at all preserve offices and work buildings. Include a copy as an Appendix to your weed control plan.

**POSTING TREATED AREAS**

Federal requirements for posting treated areas, if any, are listed on the herbicide label. Glyphosate, triclopyr and most other herbicides used in natural areas have no federal posting requirements. Some municipalities and counties have stricter requirements (e.g., Boulder, Colorado). Always keep treated areas off limits to the public at least until the herbicide dries. Treated areas may be kept off limits for longer periods if the herbicide is persistent in the environment.

When posting areas that are accessible to the public (trails, visitor centers etc.), place notices at the usual points of entry or along the perimeter of treated sites. The posting should include a notice that the area has or will be treated, the name of the herbicide used, the date of the treatment, appropriate precautions to be taken, the date when re-entry is judged to be safe, and a phone number for additional information. The notices should be removed after it is judged safe to re-enter the area.

**STORING HERBICIDES**

Store herbicides in a well ventilated, cool, dry area where food and drinks are never stored or prepared. Most pesticides should not be stored for any length of time below 40° F. The floor should be concrete or lined with plastic or other impermeable material to prevent leaks from reaching the soil.

The area should be inaccessible to the public and/or locked except when chemicals are being removed or returned. Containers should be labeled to indicate the following: contents (ratio of herbicide, surfactant, water, etc.), date mixed, and approximate volume remaining when placed in storage. The containers must be stored carefully and never stacked.

Heavy plastic garbage bags, a shovel, and a soil absorbent (e.g., cat litter) must be available for use in cleaning-up small leaks or spills. For more information on spills see below.

**MIXING HERBICIDES**

USE EXTREME CAUTION WHEN MIXING HERBICIDES! Dermal exposure to a small amount of a concentrated herbicide can be equivalent to the exposure received after a full day of working in a treated field (Libich et al. 1984). Before mixing any herbicide, READ THE LABEL. Herbicide labels are legal documents and users are obligated to read and obey them.

Establish a mixing area. Herbicides should be mixed only in pre-designated areas - preferably either in an industrial sink near the storage site or in an area near the treatment site(s) in which damage from small spills or other herbicide contamination would be minimal. Field mixing sites should have relatively few native or other desirable species, not be susceptible to erosion or runoff, and rarely, if ever, be visited by the public or preserve staff. In addition, mixing sites should provide easy access for containment and clean up of spills.

Establish a mixing area. Herbicides should be mixed only in pre-designated areas - preferably either in an industrial sink near the storage site or in an area near the treatment site(s) in which damage from small spills or other herbicide contamination would be minimal. Field mixing sites should have relatively few native or other desirable species, not be susceptible to erosion or runoff, and rarely, if ever, be visited by the public or preserve staff. In addition, mixing sites should provide easy access for containment and clean up of spills.

Prior to mixing, determine the order that chemicals will be added to the mix. Generally, adjuvants are added prior to the herbicide, but consult the label for specific instructions. When mixing, start by filling the spray tank or other mixing vessel half to three-quarters full with water. The water should be clean and clear to prevent contamination of the mixture or clogging of tank nozzles and hoses. The water should have a neutral or slightly acidic pH, as alkaline water can cause the pesticide to breakdown prior to application. Add a buffer or acidifier to the water if necessary.

Carefully measure the herbicide concentrate and add it to the tank water. Small measuring errors can lead to large errors in the amount of pesticide applied. Be aware of if you are using the active ingredient (a.i.) or acid equivalent (a.e.) of the herbicide (see sidebar below for more details). The measuring container should be rinsed and the rinsate added to the tank solution. The container of liquid herbicides should be triple rinsed with ¼ container volume of water. Add rinsate to the tank solution or store it in a separate container labeled "WATER AND RINSATE FOR HERBICIDE ONLY, NONPOTABLE"

**ACTIVE INGREDIENT (A.I.) VS. ACID EQUIVALENT (A.E.)**

Labels on herbicide containers and instructions for mixing herbicides sometimes use units of herbicide active ingredient (a.i.) or acid equivalent (a.e.). The herbicide may be sold in different concentrations, but units of a.i. or a.e. provide standard measures, so the mixing instructions can apply in all cases. In order to follow these instructions, you will need to determine how many a.i. or a.e. are in an ounce, or quart or liter, of the concentrate on hand.

The "active ingredient" (a.i.) of an herbicide formulation is responsible for its herbicidal activity or ability to kill or suppress plants. The a.i. is always identified on the herbicide label by either its common name or chemical name, or both. Herbicide formulations available for sale commonly contain other so-called "inert" compounds too.

The "acid equivalent" (a.e.) of an herbicide is just the acid portion of the a.i., and it is this acid portion that is responsible for herbicidal effects. The acid portion (or parent acid) is generally associated with other chemical compounds to form a salt or an ester, which is more stable and better able to move through a plant's waxy cuticle, and into the plant. The salt or ester is the a.i.

Weak acid herbicides are formulated as salts or esters through the addition of a salt or ester molecular group to the parent acid molecule. This allows the herbicide acid to mix properly with adjuvants and enhances the compound's ability to move into plant tissue. Once the herbicide enters the plant, the salt or ester group is cleaved off the parent molecule, allowing the acid to affect the plant.

Because the salt or ester molecular group can vary dramatically in size, a measure of the percent a.i., especially in the case of a weak acid herbicide, does not adequately reflect the percentage of acid in the formulation. Thus, the a.e. is used to determine the amount of the product to be applied.

Product labels for weak acid herbicides will list the product's percentage of active ingredient, as well as other inert ingredients, at the top of the label. The percentage of acid equivalent in the formulation is usually listed below these percentages in a separate table or paragraph.

**TRANSPORTING HERBICIDES**

Herbicides should be transported in tightly sealed containers placed in a well-constructed and watertight carrying box or bucket, such as a Rubbermaid tub or cat litter bucket. A good container will prevent leaks in vehicles, onto applicators, or to the environment. Each program should develop techniques and use materials that will best serve the needs of a particular site or circumstance. In some cases, you may want to carry only

a small amount of herbicide to treat weeds encountered while conducting daily activities in the field.

Jack McGowan-Stinski of TNC's Michigan program uses large five-gallon buckets with tight lids to transport herbicides and application equipment into the field. The buckets are large enough to hold all the necessary equipment and can be carried by groups of volunteers. Jennifer Hillmer of TNC's Ohio Program often treats weeds distributed over great distances while working in the field by herself. Jennifer keeps pesticides in a crook-necked squirt bottle for easy application and carries the squirt bottle and other application equipment in a four-liter, square, leak-proof, Nalgene bottle, which can be carried in her backpack along with other field equipment. Jennifer recommends laboratory supply companies as a good place to find equipment for herbicide application and storage.

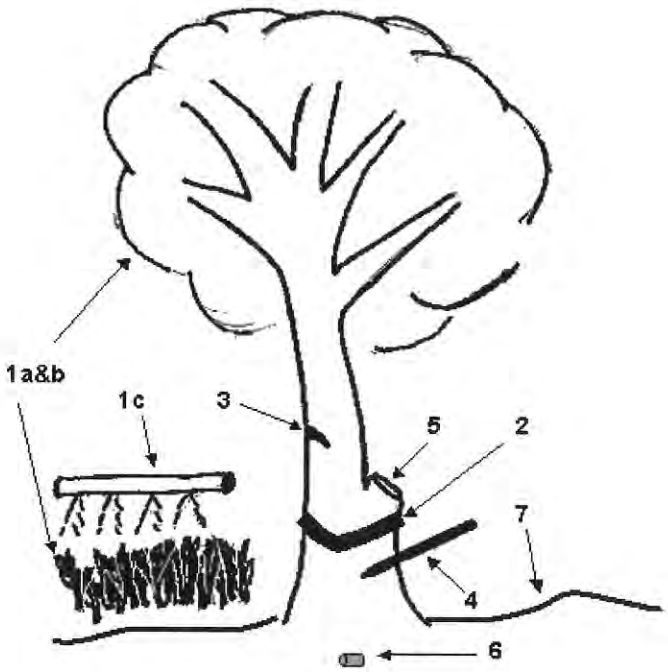
## Application of Herbicides

### Application Methods

Herbicides can be applied in a variety of ways. The most appropriate application method is determined by the weed being treated, the herbicide being applied, the skills of the applicator, and the application site. Standard application techniques can sometimes be modified to better suit the needs of natural area management. A few land managers have come up with simple but ingenious techniques and tools that save money, are more effective and safer, and are easier to use than standard methods. We include some of these in the detailed descriptions of techniques below, and encourage you to innovate because there is still plenty of room for improvement.

Methods of application (diagrammed below) can be broadly classified as follows:

1. To intact, green leaves (foliar application)
   A. Spot application (backpack applicator, spray bottle);
   B. Spot application (backpack applicator, spray bottle);
   C. Boom application
2. Around the circumference of the trunk on the intact bark (basal bark);
3. Around the circumference of the trunk on the intact bark (basal bark);
4. To cut stems and stumps (cut stump);
5. In pellet form at the plant's base (rarely used in natural areas);
6. To the soil before the target species seeds germinate and emerge (rarely used in natural areas).




1. Foliar Applications
   These methods apply herbicide directly to the leaves and stems of a plant. An adjuvant or surfactant is often needed to enable the herbicide to penetrate the plant cuticle, a thick, waxy layer present on leaves and stems of most plants. There are several types of foliar application tools available.
   A. Spot applicators - Spray herbicide directly onto target plants only, and avoid spraying other desirable plants. These applicators range from motorized rigs with spray hoses to backpack sprayers, to hand-pumped spray or squirt bottles, which can target very small plants or parts of plants. Crook-necked squirt bottles and similar equipment can be ordered from laboratory supply companies and are easy to carry over distances and through dense vegetation.
   B. B. Wick (wipe-on) applicators - Use a sponge or wick on a long handle to wipe herbicide onto foliage and stems. Use of a wick eliminates the possibility of spray drift or droplets falling on non-target plants. However, herbicide can drip or dribble from some wicks.
      i. "Paint sticks" and "stain sticks" sold at local hardware stores have been used successfully for wick application. These sticks

have a reservoir in the handle that can hold herbicide, which soaks a roller brush at the end of the handle. The brush is wiped or rolled across leaves and stems.

ii. The "glove of death" is a technique developed by TNC land stewards for applying herbicide in an otherwise high quality site. Herbicide is sprayed directly onto a heavy cotton glove worn over a thick rubber/latex (or nitrile) glove. The wearer of the glove can then apply the herbicide with total precision and little or no runoff.

C. Boom applicator - A boom, a long horizontal tube with multiple spray heads, is mounted or attached to a tractor, ATV (or other four-wheel drive vehicle), helicopter, or small plane. The boom is then carried above the weeds while spraying herbicide, allowing large areas to be treated rapidly with each sweep of the boom. Offsite movement due to vaporization or drift and possible treatment of non-target plants can be of concern when using this method.

2. Basal Bark

This method applies a 6 to 12 inch band of herbicide around the circumference of the trunk of the target plant, approximately one foot above ground. The width of the sprayed band depends on the size of the plant and the species' susceptibility to the herbicide. The herbicide can be applied with a backpack sprayer, hand-held bottle, or a wick. Ester formulations are usually best for basal bark treatments, as esters can pass most readily through the bark (as compared to salts). Esters can be highly volatile, however, so basal bark treatments should be performed only on calm, cool days. During summer, treatment is best carried out in the mornings, which tend to be cooler. The basal bark treatment works best on young trees with smooth bark. It is usually not effective against older plants with thick corky bark.

3. Frill or Hack & Squirt

The frill method, also called the "hack and squirt" treatment, is often used to treat woody species with large, thick trunks. The tree is cut using a sharp knife, saw, or ax, or drilled with a power drill or other device. Herbicide is then immediately applied to the cut with a backpack sprayer, squirt bottle, syringe, or similar equipment. Because the herbicide is placed directly onto the thin layer of growing tissue in the trunk (the cambium), an ester formulation is not required.

Jack McGowan-Stinski (TNC-Michigan) recommends using the drill treatment rather than cutting, for trees with dbh (diameter at breast height) greater than three inches. He has volunteers use "tree steps" to drill holes into trees. Tree steps are large metal screws that can be screwed into a tree trunk by hand to provide steps for tree climbing. When applying herbicide, tree steps are lightweight drilling tools that can be easily carried into the field and used by untrained volunteers. These tools are available at most hunting stores and cost only a few dollars each.

Jack recommends drilling one hole for each inch in dbh. (A ten-inch dbh tree would require at least ten holes.) Holes should be drilled at a slight downward angle to prevent the herbicide from running out, and should be deep enough to penetrate the inner bark or growing tissue.

Some added recommendations made by Jack for using the drill method include: 1) Spray-paint tree steps with a neon color to prevent them from being lost if dropped in dense vegetation. 2) Spray-paint circles directly onto the trees around the drilled holes. This will ensure that no holes are overlooked by the herbicide applicator. After the hole is filled with herbicide, the applicator can spray paint a line through the hole to indicate that it was treated.

4. Injection

Herbicide pellets can be injected into the trunk of a tree using a specialized tool such as the EZ-Ject Lance. The EZ-Ject lance's five ft long, metal tube has "teeth" on one end that grip the trunk of the tree. A sharp push on the other end of the tube sends a brass capsule of herbicide into the tree trunk. It is a convenient way of applying herbicide and requires minimal preparation or clean up. In addition, it is an easy and safe way to apply herbicides with minimal exposure.

There are however, some serious drawbacks to this method. The lance and capsules are expensive ($425 per lance; approximately $500 per 4,800 capsules, depending on herbicide), and full-sized lances can be unwieldy, particularly in thickets. The lance furthermore, is difficult to thrust with enough power to drive the capsules far enough into thick barked trees to be effective. A large number of capsules placed close together are often necessary to kill large trees.

At the Albany Pine Bush Preserve in New York, glyphosate gel pellets were injected using an EZ-Ject Lance into trees with an average dbh of eight centimeters. In some cases, crowns of treated trees later showed signs of stress, but most of these re-sprouted vigorously and none of the treated trees died (Hawver et al. 2000).

For information or to order an EZ-Ject Lance contact Odom Processing Engineering Consulting, Inc., 800 Odom Industries Road, Waynesboro, MS, 39367, (601) 735-2680, (888) 395-6732, www.ezject.com.

Herbicides can also be injected into herbaceous stems by using a needle and syringe. Jonathan Soll (TNC-Oregon) reports 100% control of small patches of Japanese knotweed (Polygonum cuspidatum) with no off-target effects, by injecting every single stem near the base with herbicide. He adds that this method may actually use more herbicide than foliar spraying (since you use high concentations of the herbicide), and caution with the needle and syringe is necessary since you are carrying around a sharp object.

5. Cut-Stump

This method is often used on woody species that normally re-sprout after being cut. Cut down the tree or shrub, and immediately spray or squirt herbicide on the exposed cambium (living inner bark) of the stump. The herbicide must be applied to the entire inner bark (cambium) within minutes after the trunk is cut. The outer bark and heartwood do not need to be treated since these tissues are not alive, although they support and protect the tree's living tissues.

Herbicide can be applied to cut stumps in many ways, including spray and squirt bottles, or even paint brushes. Care must be taken to avoid applying too much herbicide, and allowing it to run-off the stump and onto the ground. Herbicide can also dribble from bottles or brushes and fall on desirable plants or the ground. To help avoid these problems, Jack McGowan-Stinski (TNC-Michigan) developed an inexpensive and easy to assemble application tool using PVC pipe and a sponge through which the herbicide can be applied. See the Appendix for a diagram and instructions on how to build one.

Sometimes even treated stumps will re-sprout, so it is important to check them at regular intervals (2 to 6 months) for at least a year. Depending on the vigor of the re-sprouts, these can be treated by cutting, basal bark applications, or foliar applications. Even when foliar

applications are called for, treating re-sprouts is usually far easier and requires much less herbicide than treating the tree (before it was cut down) with a foliar application.

The cut stump treatment allows for a great deal of control over the site of herbicide application, and therefore, has a low probability of affecting non-target species or contaminating the environment. It also requires only a small amount of herbicide to be effective. Black locust (Robinia pseudoacacia) and buckthorns (Rhamnus spp.) have been successfully controlled using this method (Hawver et al. 2000; J. McGowan-Stinski, pers. comm.).

**Selecting a Method**
**Minimize**

Select a technique(s) that (1) minimizes risks of contact to the applicator and others that may be in the area during and after herbicide application, AND (2) minimizes release of herbicide to the environment, particularly if the herbicide could contact non-target species. Avoid using boom application where possible (1c above) because it can result in a relatively high amount of herbicide contacting non-target species and bare ground. Also, avoid using pellets and pre-emergence herbicides (6 & 7 above, respectively) because they are relatively persistent in the environment.

**Use a dye**

Mix a dye with the herbicide so applicators can see which plants have been treated and if they have gotten any herbicide on themselves or their equipment. Some pre-mixed herbicides include a dye (e.g., Pathfinder II® includes the active ingredient triclopyr, a surfactant, and a dye). Ester based herbicides like Garlon 4® require oil-soluble dyes like colorfast purple®, colorfast red®, and basoil red® (for use in basal bark treatments), which are sold by agricultural chemical and forestry supply companies. Clothing dyes like those produced by Rit® will work in water-soluble herbicides such as Garlon 3A®. These dyes are inexpensive and available at most supermarkets and drugstores.

**Who May Apply Herbicides?**

TNC employees or contractors who apply herbicides must have all certificates or licenses required by the state. Each state has its own requirements. Some require applicators working in natural areas to be certified and others do only if compounds designated "restricted-use" by the EPA or the state are to be used. Most states conduct applicator training programs and in many areas local Agricultural Extension Agents give workshops on proper herbicide use.

Volunteers may NOT apply herbicides unless they are properly licensed AND have signed a consent & release form. An example of such a form produced by the Illinois Field Office is provided as an Appendix. **Check with the legal staff for your program before drafting one of these forms or using volunteers to apply herbicides**. TNC staff who supervise volunteers should be properly licensed or certified.

**Protection Against Herbicides**

When using herbicides, the safety of the applicator, to others, and to the environment is of utmost importance. Be sure to read the earlier textbox (page 5.6) on "Personal Protection in Herbicide Handling" regarding specific equipment requirements, how to avoid contamination, eye and respiratory protection, how to clean-up after herbicide use, and how to launder clothes and equipment used during herbicide application.

**When to Apply Herbicides**

The best time to apply an herbicide is determined primarily by the herbicide's mode of action and the physiology of the target plants. In seasonal climates, it is often best to apply herbicides in autumn or prior to the dry season, 3 to 6 weeks before the target plant goes dormant for the season. This is because many plants apparently transfer sugars and nutrients from their stems and leaves to belowground storage organs at this time and will carry herbicides along to these areas as well. Contrary to assumptions that plants will be most vulnerable when weak, herbicides are usually ineffective when applied during a drought or other stressful conditions. This is because most herbicides work by attacking growing tissue and metabolic processes, which plants 'shut down' when stressed. In fact, late winter or early spring are often good times to apply herbicide because this is when plants begin growing again, and can efficiently translocate the herbicide throughout their tissues. Fosamine ammonium, the dormancy enforcer, is best applied in the late fall just before leaf drop. The herbicidal effects of fosamine ammonium however, are not observed until the following spring when treated plants fail to re-foliate.

In some cases, the site of application may determine the best time to apply a herbicide. For example, buckthorns (Rhamnus spp.) growing in wet, boggy areas are easiest to treat during winter when the ground is frozen. Check the label or consult your distributor for the best application time under the conditions at your site.

Note that with some herbicides there is a long time lag between time of herbicide application and the first evidence that they are working. This is particularly true of herbicides that work by inhibiting amino acid or lipid synthesis, because the plant(s) can rely on stored supplies to continue growing.

**Record Keeping**

When using herbicides it is critical (and, in some cases, required by law) to keep records of all plants/areas treated, amounts and types of herbicide used, and dates of application. This information will be important in evaluating the project's success, improving methodology, and identifying mistakes. In addition, it documents the procedure for future site managers and biologists. Records of abundance/condition of the targeted weeds and nearby desirable plants before and after treatment will also be valuable in evaluating the effectiveness of the herbicide.

## Herbicide Disposal

**Equipment cleanup**

Following use, application equipment and empty containers should be triple rinsed with clean water using 10% of the container volume for each rinse. If possible, rinse equipment in the treatment area and apply the wastewater to weeds or store for future use as a dilutant. Left over herbicide mix that will not be used later should be treated as hazardous waste.

**Container disposal**

Use the state herbicide container recycling program where available. In Minnesota, herbicide dealers are required to collect empty containers from customers. If no specific agri-chemical container recycling program is available, puncture the empty container to prevent anyone from using it as a container again, and then dispose of or destroy it. In most areas, small numbers of empty, triple-rinsed containers can be disposed in the trash for pick-up or taken to the local dump, unless the label states otherwise. In parts of California and some other states you may be required to get written permission from your County Agriculture Commissioner to dispose of containers. Call your local Commissioner for details. Some jurisdictions require containers to be burned, while others prohibit burning pesticide containers. If the herbicide label states that the container may not be disposed of in regular sanitary landfills, call your county or municipal waste department for information on Hazardous Material Collection dates.

**Equipment and applicator clean-up**

After use, first clean and store application equipment and then thoroughly rinse personal protection gear (gloves, boots, etc.) with cold water from a hose or container that is hand-held (gloves off) and was not used during application work. All personal protection gear should then be washed in mild soap and water. Finally, applicators should wash their hands and any herbicide-exposed areas of their bodies. Applicators should shower and change clothing as soon as possible. Clothes used during the application must be washed and dried separately from other clothing before it is worn again, even if it appears uncontaminated.

**Contaminated clothing**

If herbicide concentrate spills on clothing, the clothing should be discarded or, where permitted, burned immediately. Wrap contaminated clothing and other materials in newspaper before placing in trash or landfill. Clothing and other items contaminated with certain commercial products, such as technical grade 2,4-D or formulations in which 2,4-D is the only active ingredient, are classed as hazardous waste. Call your local hazardous materials center for instructions on how to dispose of this material. In cases where small quantities are involved it may be possible to dispose of contaminated clothing in the trash.

## Responding to Spills

Rules and regulations regarding pesticide spills vary between states and counties. Therefore, before obtaining herbicides, call the local fire department or county Hazardous Materials Office for information on local regulations. In most cases, the proper response to a spill depends on the volume and concentration of herbicide released, location of the spill, and the chemical(s) involved. If possible, inquire as to whether a report would be required in a hypothetical situation in which all the herbicide was spilled (1) on the soil in the interior of the preserve and (2) along a public road. A rule of thumb employed by some public land management agencies is not to call for help from the local Hazardous Materials Office for herbicide spills unless they contaminate too much soil to dig up and place in plastic garbage bags. However, since our goal is to protect biodiversity, land managers are expected to minimize damage to native populations. Hazardous Materials officers we spoke to considered spills under 100 gallons to be "small". Most emergency systems appear to be designed to deal with these larger volumes used in agriculture and industry, which are far larger than those typically used in natural areas.

Be sure to carry a "Pesticide Kit" for emergency spills (see the following Pesticide Spill Kit equipment list). If a spill occurs, keep people away from affected areas until the clean-up process is complete. When small volumes of dilute herbicide are spilled they may be treated by carefully digging up the affected soil and litter, and spreading this material at the legal rate or concentration. Small diesel (sometimes used as a crude surfactant) and gasoline spills may be treated by adding organic material (e.g., cow manure or compost) to the affected area and keeping it moist. It may take several years for the spilled material to degrade.

**PESTICIDE KIT EQUIPMENT LISTS**

adapted from work by Jack McGowan-Stinski and Jennifer Hillmer

**PESTICIDE SPILL KITS**

- Emergency phone numbers
- Personal Protective Equipment: gloves, footwear, apron, goggles, face shield, respirator
- Heavy plastic bags for material storage
- Containment "snakes" (chemsorb tubes or pads to contain & absorb spilled chemicals)
- Absorbent materials (cat litter, vermiculite, paper, etc.)
- Neutralizing agents (bleach and hydrated lime)
- Sweeping compound for dry spills
- Shovel, broom, dustpan
- Heavy duty detergent, chlorine bleach, and water
- Fire extinguisher certified for all types of fires
- Sturdy plastic container that closes tightly and will hold the largest quantity of pesticide on hand
- First aid supplies
- Fresh water (at least 3 gallons; bring extra for wash-up after application)

- Eyewash
- Soap (dish soap or hand soap)
- Towels
- Change of clothes
- Additional items required by labeling

**ADDITIONAL HERBICIDE FIELD EQUIPMENT**

- Extra application equipment (e.g., squeeze bottles, nalgene bottles, sponges)
- Funnel
- Herbicide dyes
- Herbicide in original containers
- Extra water, soap, towels, plastic bags

In any spill considered to be an emergency, call the local fire department. They may come to the site to help prevent further spread of the chemical but if the spill is large they will likely require a certified company to do the clean-up.

Companies often charge initial fees of roughly $2,000 plus hourly fees of $100/hour for the work to meet minimum legal clean-up requirements. If a spill occurs and there is uncertainty about legal requirements for reporting and clean-up, contact the program's legal staff immediately. They can ensure that all federal, state and local regulations are met.

**REFERENCES**

Hawver, C. A., S. B. Gifford, and J. Hecht. 2000. Comparison of methods to control invasive plant species in the Albany Pine Bush Preserve. Albany Pine Bush Preserve Commission, Latham, New York. 50 pgs.
Libich, S., J. C. To, R. Frank, and G. J. Sirons. 1984. Occupational exposure of herbicide applicators of herbicides used along electric power transmission line right-of-way. Am. Ind. Hyg. Assoc. J. 45(1):56-62.

**Date Authored**: April 2001
**Updated**: June 2003

**Herbicide Properties**

Herbicides belong to a group of chemicals known as pesticides, which prevent, destroy, repel, or mitigate any pest. Herbicides are any chemical substance that is used to specifically kill plants. Other familiar pesticides are insecticides, rodenticides, and fungicides.

MODE OF ACTION

An herbicide's mode of action is the biochemical or physical mechanism by which it kills plants. Most herbicides kill plants by disrupting or altering one or more of a their metabolic processes. Some disrupt the cellular membranes of plants, allowing cellular contents to leak out, but do not directly disrupt other metabolic processes. Some species or whole groups of plants are not susceptible to certain herbicides because they use different biochemical pathways or have slightly different enzymes. Animals typically suffer little or no effect from most herbicides sold today because these compounds principally affect processes exclusive to plants, like photosynthesis or production of aliphatic amino acids.

**HERBICIDE FAMILIES VS. MODE OF ACTION**

Herbicides that are chemically similar are said to belong to the same "herbicide family". The compounds in a given family typically exhibit similar characteristics and function, due to their chemical and structural similarities. For example, clopyralid, picloram, and triclopyr are all grouped in the pyridine family.

An herbicide's mode of action is the mechanism (biochemical or physical) by which it kills or suppresses plants. The mode of action is generally dictated by its chemical structure, and therefore, herbicides in the same family, tend to have the same Mode of Action. For instance, clopyralid, picloram, and triclopyr are all in the pyridine family and are all auxin mimic herbicides, while glyphosate is an amino acid inhibitor. Some herbicides from different families, however, can have the same mode of action. For example, the phenoxy 2,4-D is an auxin mimic, just like the pyridines picloram, clopyralid, and triclopyr.

The Herbicide Table in this handbook indicates the family and mode of action for each herbicide covered in this manual.

An herbicide is often chosen for use based on its mode of action. If one herbicide is ineffective, another herbicide with a different mode of action may provide better results. When and how an herbicide is applied may be determined by its mode of action.

"Pre-emergent" herbicides are those applied to the soil before the weed germinates, and either disrupt germination or kill the germinating seedling. "Post-emergent" herbicides are those that are applied directly to already established plants and/or soil. Some herbicides are effective both before ("pre-emergent") and after ("post-emergent") germination.

Some of the most common modes of action include:

- Auxin mimics (2,4-D, clopyralid, picloram, and triclopyr), which mimic the plant growth hormone auxin causing uncontrolled and disorganized growth in susceptible plant species;
- Mitosis inhibitors (fosamine), which prevent re-budding in spring and new growth in summer (also known as dormancy enforcers);
- Photosynthesis inhibitors (hexazinone), which block specific reactions in photosynthesis leading to cell breakdown;
- Amino acid synthesis inhibitors (glyphosate, imazapyr and imazapic), which prevent the synthesis of amino acids required for construction of proteins;
- Lipid biosynthesis inhibitors (fluazifop-p-butyl and sethoxydim), that prevent the synthesis of lipids required for growth and maintenance of cell membranes.

**Auxin Mimics**

Picloram, clopyralid, triclopyr, and 2,4-D are referred to as synthetic auxins. Auxin is a plant hormone that regulates growth in many plant tissues. Chemically, 2,4-D is classified as a phenoxy acetic acid, while picloram, clopyralid, and triclopyr are pyridine derivatives. When susceptible plants are treated with these herbicides, they exhibit symptoms that could be called 'auxin overdose', and eventually die as a result of increased rates of disorganized and uncontrolled growth.

In use since 1945, 2,4-D is one of the most studied herbicides in the world. It is known to affect many biochemical processes in plants, but it is still not clear which of the biochemical alterations 2,4-D and other auxin-mimic herbicides cause that is ultimately responsible for killing plants. It is possible that plants are weakened more or less equally by several of these disruptions with no one process being the most important.

The sequence of events following treatment with an auxin mimic herbicide differs from one species to another and depends on the age and physiological state of the individual plant. Marked changes in the permeability of the plant's cell wall or membrane can generally be detected within minutes of application. This change may lead to a rapid and sustained loss of H+ ions (protons) from the cell wall, which makes the wall more elastic, and often results in measurable cell growth within an hour. The loss of H+ ions may also lead to an accumulation of K+ ions in the stomatal guard cells, causing those cells to swell, increasing the size of the stomatal opening. The increased stomatal opening helps bring about a short-lived increase in photosynthesis, presumably because it allows higher concentrations of $CO_2$ to reach the photosynthesizing cells inside the leaf.

Other biochemical changes that occur within a day of treatment include a large increase in the concentration of soluble sugars and amino acids inside cells. This increase coincides with an increase in messenger RNA synthesis and a large increase in rates of protein synthesis. Treated plants also frequently produce ethylene, a gaseous plant hormone.

Grasses and other monocots are generally not susceptible to auxin-mimic herbicides. The reason for this selectivity is unclear because there are no apparent differences between the binding sites targeted by auxins in monocots and dicots. It may, however, be due to differences in vascular tissue structure or differences in ability to translocate or metabolize the herbicide (DiTomaso 1999).

**Mitosis Inhibitors**

Fosamine ammonium is another herbicide that acts as a plant growth regulator. It is sometimes referred to as a "dormancy enforcer," but the specific mechanism of action has not been identified, even though there is evidence that fosamine ammonium inhibits mitosis in susceptible plants. When applied to deciduous plants up to two months before leaf drop, the compound is absorbed with little or no apparent effect. The following spring however, the plants fail to leaf-out because bud development is either prevented or limited to spindly, miniature leaves. Plants often die as the season progresses because they cannot produce enough photosynthate to sustain themselves. A distinctive feature of this mode of action is that treated plants do not go through a "brown-out" phase, as is often seen after the application of other herbicides. Susceptible non-deciduous plants such as pines, die soon after application because they simply do not produce enough photosynthate.

**Photosynthesis Inhibitors**

There are two types of photosynthesis inhibitors. Hexazinone is an example of the type that inhibits the transfer of electrons in photosystem II. It blocks electron transport from QA to QB in the chloroplast thylakoid membranes by binding to the D-1 protein at the QB-binding niche. The electrons blocked from passing through photosystem II are transferred through a series of reactions to other reactive toxic compounds. These compounds disrupt cell membranes and cause chloroplast swelling, membrane leakage, and ultimately cellular destruction.

Paraquat and diquat are examples of the second type of photosynthesis inhibitor. They accept electrons from Photosystem I, and after several cycles, generate hydroxyl radicals. These radicals are extremely reactive and readily destroy unsaturated lipids, including membrane fatty acids and chlorophyll. This destroys cell membrane integrity, so that cells and organelles "leak", leading to rapid leaf wilting and desiccation, and eventually to plant death (WSSA 1994).

**Amino Acid Synthesis Inhibitors**

Picloram, clopyralid, triclopyr, and 2,4-D are referred to as synthetic auxins. Auxin is a plant hormone that regulates growth in many plant tissues. Chemically, 2,4-D is classified as a phenoxy acetic acid, while picloram, clopyralid, and triclopyr are pyridine derivatives. When susceptible plants are treated with these herbicides, they exhibit symptoms that could be called 'auxin overdose', and eventually die as a result of increased rates of disorganized and uncontrolled growth.

In use since 1945, 2,4-D is one of the most studied herbicides in the world. It is known to affect many biochemical processes in plants, but it is still not clear which of the biochemical alterations 2,4-D and other auxin-mimic herbicides cause that is ultimately responsible for killing plants. It is possible that plants are weakened more or less equally by several of these disruptions with no one process being the most important.

The sequence of events following treatment with an auxin mimic herbicide differs from one species to another and depends on the age and physiological state of the individual plant. Marked changes in the permeability of the plant's cell wall or membrane can generally be detected within minutes of application. This change may lead to a rapid and sustained loss of H+ ions (protons) from the cell wall, which makes the wall more elastic, and often results in measurable cell growth within an hour. The loss of H+ ions may also lead to an accumulation of K+ ions in the stomatal guard cells, causing those cells to swell, increasing the size of the stomatal opening. The increased stomatal opening helps bring about a short-lived increase in photosynthesis, presumably because it allows higher concentrations of CO2 to reach the photosynthesizing cells inside the leaf.

Other biochemical changes that occur within a day of treatment include a large increase in the concentration of soluble sugars and amino acids inside cells. This increase coincides with an increase in messenger RNA synthesis and a large increase in rates of protein synthesis. Treated plants also frequently produce ethylene, a gaseous plant hormone.

Grasses and other monocots are generally not susceptible to auxin-mimic herbicides. The reason for this selectivity is unclear because there are no apparent differences between the binding sites targeted by auxins in monocots and dicots. It may, however, be due to differences in vascular tissue structure or differences in ability to translocate or metabolize the herbicide (DiTomaso 1999).

**Mitosis Inhibitors**

Fosamine ammonium is another herbicide that acts as a plant growth regulator. It is sometimes referred to as a "dormancy enforcer," but the specific mechanism of action has not been identified, even though there is evidence that fosamine ammonium inhibits mitosis in susceptible plants. When applied to deciduous plants up to two months before leaf drop, the compound is absorbed with little or no apparent effect. The following spring however, the plants fail to leaf-out because bud development is either prevented or limited to spindly, miniature leaves. Plants often die as the season progresses because they cannot produce enough photosynthate to sustain themselves. A distinctive feature of this mode of action is that treated plants do not go through a "brown-out" phase, as is often seen after the application of other herbicides. Susceptible non-deciduous plants such as pines, die soon after application because they simply do not produce enough photosynthate.

**Photosynthesis Inhibitors**

There are two types of photosynthesis inhibitors. Hexazinone is an example of the type that inhibits the transfer of electrons in photosystem II. It blocks electron transport from QA to QB in the chloroplast thylakoid membranes by binding to the D-1 protein at the QB-binding niche. The electrons blocked from passing through photosystem II are transferred through a series of reactions to other reactive toxic compounds. These compounds disrupt cell membranes and cause chloroplast swelling, membrane leakage, and ultimately cellular destruction.

Paraquat and diquat are examples of the second type of photosynthesis inhibitor. They accept electrons from Photosystem I, and after several cycles, generate hydroxyl radicals. These radicals are extremely reactive and readily destroy unsaturated lipids, including membrane fatty acids and chlorophyll. This destroys cell membrane integrity, so that cells and organelles "leak", leading to rapid leaf wilting and desiccation, and eventually to plant death (WSSA 1994).

**Amino Acid Synthesis Inhibitors**

Glyphosate and imazapyr kill plants by preventing the synthesis of certain amino acids produced by plants but not animals. Glyphosate blocks the action of the enzyme 5-enolpyruvylshikimate-3-phosphate (EPSP) synthase, which inhibits the biosynthesis of certain aromatic amino acids such as phenylalanine, tyrosine, and tryptophan. These amino acids are required for protein synthesis, which, in turn, is necessary for plant growth and maintenance. Other biochemical processes such as carbohydrate translocation, can also be affected by these herbicides. Although these effects are considered secondary, they may be important in the total lethal action of glyphosate.

Imazapyr, another amino acid synthesis inhibitor, kills plants by inhibiting the production of the branched-chain aliphatic amino acids (valine, leucine, and isoleucine), which are required for DNA synthesis and cell growth. It does this by blocking acetohydroxy acid synthase (AHAS), also known as acetolactate synthase (ALS). Plants treated with imazapyr usually die slowly. The time it takes for a treated plant to die is most likely related to the amount of stored aliphatic amino acids available to the plant. AHAS (ALS) is widespread in plants but the biochemical pathway it catalyzes is not found in animals.

**Lipid Biosynthesis Inhibitors**

Fluazifop-p-butyl and sethoxydim are both grass specific herbicides that inhibit the synthesis of enzymes required for lipid synthesis. Both inhibit acetyl CoA carboxylase, the enzyme responsible for catalyzing an early step in fatty acid synthesis. Non-susceptible broadleaf species have a different binding site, rendering them immune. The inhibition of acetyl CoA carboxylase and the subsequent lack of lipid production leads to losses in cell membrane integrity, especially in regions of active growth such as meristems. Eventually shoot and rhizome growth ceases, and shoot meristems and rhizome buds begin to die back.

**FORMULATIONS**

A herbicide formulation is the total marketed product, and is typically available in forms that can be sprayed on as liquids or applied as dry solids. It includes the active ingredient(s), any additives that enhance herbicide effectiveness, stability, or ease of application such as surfactants and other adjuvants, and any other ingredients including solvents, carriers, or dyes. The application method and species to be treated will determine which formulation is best to use. In most cases, manufacturers produce formulations that make applications and handling simpler and safer. Some herbicides are available in forms that can reduce risk of exposure during mixing, such as pre-measured packets that dissolve in water, or as a liquid form already mixed with surfactant and dye (e.g., Pathfinder II®).

**Sprayable/liquid formulations include:**

1. Water-soluble formulations: soluble liquids (SL), soluble powders or packets (SP), and soluble granules (SG). Only a few herbicidal active ingredients readily dissolve in water. These products will not settle out or separate when mixed with water.
2. Emulsifiable formulations (oily liquids): emulsifiable concentrates (E or EC) and gels (GL). These products tend to be easy to handle and store, require little agitation, and will not settle out of solution. Disadvantages of these products are that most can be easily absorbed through the skin and the solvents they contain can cause the rubber and plastic parts of application equipment to deteriorate.
3. Liquid suspensions (L for liquid or F for flowable) that are dispersed in water include: suspension concentrates (SC), aqueous suspensions (AS), emulsions of water-dissolved herbicide in oil (EO), emulsions of an oil-dissolved herbicide in water (EW), micro-encapsulated formulations (ME), and capsule suspensions (CS). All these products consist of a particulate or liquid droplet active ingredient suspended in a liquid. They are easy to handle and apply, and rarely clog nozzles. However, they can require agitation to keep the active ingredients from separating out.
4. Dry solids that are suspended in water: wettable powders (W or WP), water-dispersible granules (WDG, WG, DG), or dry flowables (DF). These formulations are some of the most widely used. The active ingredient is mixed with a fine particulate carrier, such as clay, to maintain suspension in water. These products tend to be inexpensive, easy to store, and are not as readily absorbed through the skin and eyes as ECs or other liquid formulations. These products, however, can be inhalation hazards during pouring and mixing. In addition, they require constant agitation to maintain suspension and they may be abrasive to application pumps and nozzles.

**Dry formulations include:**

1. Granules (G) - Granules consist of the active ingredient absorbed onto coarse particles of clay or other substance, and are most often used in soil applications. These formulations can persist for some time and may need to be incorporated into the soil.
2. Pellets (P) or tablets (TB) - Pellets are similar to granules but tend to be more uniform in size and shape.
3. Dusts (D) - A dust is a finely ground pesticide combined with an inert or inactive dry carrier. They can pose a drift or inhalation hazard.

**Salts vs. Esters**

Many herbicidally active compounds are acids that can be formulated as a salt or an ester for application. Once the compound enters the plant, the salt or ester cation is cleaved off allowing the parent acid (active ingredient) to be transported throughout the plant. When choosing between the salt or ester formulation, consider the following characteristics:

**Salts**

- Most salts are highly water soluble, which reduces the need for emulsifiers or agitation to keep the compound suspended.
- Salts are not soluble in oil.
- Salts generally require a surfactant to facilitate penetration through the plant cuticle (waxy covering of leaves and stems).
- Salts are less volatile than esters.
- Salts can dissociate in water. In hard water the parent acid (i.e. the active ingredient) may bind with calcium and magnesium in the water, precipitate out, and be inactivated.

**Esters**

- Esters can penetrate plant tissues more readily than salts, especially woody tissue
- Esters generally are more toxic to plants than salts
- Esters are not water soluble and require an emulsifying agent to remain suspended in water-based solvents
- Esters have varying degrees of volatility

**Adjuvants (including surfactants)**

An adjuvant is any material added to a pesticide mixture that facilitates mixing, application, or pesticide efficacy. An adjuvant enables an applicator to customize a formulation to be most effective in a particular situation. Adjuvants include surfactants, stickers, extenders, activators, compatibility agents, buffers and acidifiers, deposition aids, de-foaming agents, thickeners, and dyes. See the Adjuvant Chapter (Chapter 8) in this handbook for more details on adjuvants.

**Surfactants**

Surfactants are the most important adjuvants. They are chemical compounds that facilitate the movement of the active herbicide ingredient into the plant. They may contain varying amounts of fatty acids that are capable of binding to two types of surfaces, such as oil and water. Some herbicide formulations come with a surfactant already added, in others, surfactants can be added prior to application. Whether a surfactant should be added will be determined by the type of herbicide being applied and the target plant. Read the label for recommendations of appropriate surfactants.

## Mechanisms of Dissipation

Dissipation refers to the movement, degradation, or immobilization of an herbicide in the environment.

**Degradation**

Degradation occurs when an herbicide is decomposed to smaller component compounds, and eventually to $CO_2$, water, and salts through

photochemical, chemical, or biological (microbial metabolism) reactions (Freed and Chiou 1981). Biodegradation accounts for the greatest percentage of degradation for most herbicides (Freed and Chiou 1981). When a single herbicide degrades, it usually yields several compounds ("metabolites"), each of which has its own chemical properties including toxicity, adsorption capacity, and resistance to degradation. Some metabolites are more toxic and/or persistent than the parent compound. In most cases, the nature of the metabolites are largely unknown.

**Photodegradation**

Photodegradation refers to decomposition by sunlight. Sunlight intensity varies with numerous factors including latitude, season, time of day, weather, pollution, and shading by soil, plants, litter, etc. Studies of the photodegradation of herbicides are often conducted using UV light exclusively, but there is some debate as to whether most UV light actually reaches the surface of the earth. Therefore, photodegradation rates determined in the laboratory may over-estimate the importance of this process in the field (Helling et al. 1971).

**Microbial Degradation**

Microbial degradation is decomposition through microbial metabolism. Different microbes can degrade different herbicides, and consequently, the rate of microbial degradation depends on the microbial community present in a given situation (Voos and Groffman 1997, McCall et al. 1981). Soil conditions that maximize microbial degradation include warmth, moisture, and high organic content.

Herbicides may be microbially degraded via one of two routes. They may be metabolized directly when they serve as a source of carbon and energy (i.e. food) for microorganisms (Hutzinger 1981), or they may be co-metabolized in conjunction with a naturally occurring food source that supports the microbes (Hutzinger 1981). Herbicides that are co-metabolized do not provide enough energy and/or carbon to support the full rate of microbial metabolism on their own.

There is sometimes a lag time before microbial degradation proceeds. This may be because the populations of appropriate microbes or their supplies of necessary enzymes start small, and take time to build-up (Farmer and Aochi 1987, Kearney and Karns 1960). If this lag time is long, other degradation processes may play more important roles in dissipation of the herbicide (Farmer and Aochi 1987). Degradation rates of co-metabolized herbicides tend to remain constant over time.

**Chemical Decomposition**

Chemical decomposition is degradation driven by chemical reactions, including hydrolyzation (reaction with hydrogen, usually in the form of water), oxidation (reaction with oxygen), and disassociation (loss of an ammonium or other chemical group from the parent molecule). The importance of these chemical reactions for herbicide degradation in the field is not clear (Helling et al. 1971).

**Immobilization/Adsorption**

Herbicides may be immobilized by adsorption to soil particles or uptake by non-susceptible plants. These processes isolate the herbicide and prevent it from moving in the environment, but both adsorption and uptake are reversible. In addition, adsorption can slow or prevent degradation mechanisms that permanently degrade the herbicide.

Adsorption refers to the binding of herbicide by soil particles, and rates are influenced by characteristics of the soil and of the herbicide. Adsorption is often dependent on the soil or water pH, which then determines the chemical structure of the herbicide in the environment. Adsorption generally increases with increasing soil organic content, clay content, and cation exchange capacity, and it decreases with increasing pH and temperature. Soil organic content is thought to be the best determinant of herbicide adsorption rates (Farmer and Aochi 1987, Que Hee and Sutherland 1981, Helling et al. 1971). Adsorption is also related to the water solubility of an herbicide, with less soluble herbicides being more strongly adsorbed to soil particles (Helling et al. 1971). Solubility of herbicides in water generally decreases from salt to acid to ester formulations, but there are some exceptions. For example, glyphosate is highly water-soluble and has a strong adsorption capacity.

The availability of an herbicide for transport through the environment or for degradation is determined primarily by the adsorption/desorption process (WHO 1984). Adsorption to soil particles can stop or slow the rate of microbial metabolism significantly. In other cases, adsorption can facilitate chemical or biological degradation (Farmer and Aochi 1987). Adsorption can change with time and, in most cases, is reversible (i.e. the herbicide can desorb from the soil or sediments and return to the soil solution or water column).

**Movement/Volatilization**

Movement through the environment occurs when herbicides are suspended in surface or subsurface runoff, volatilized during or after application, evaporated from soil and plant surfaces, or leached down into the soil. Although generally studied and discussed separately, these processes actually occur simultaneously and continuously in the environment (Hutzinger 1981).

Volatilization occurs as the herbicide passes into the gaseous phase and moves about on the breeze. Volatilization most often occurs during application, but also can occur after the herbicide has been deposited on plants or the soil surface. The volatility of an herbicide is determined primarily by its molecular weight. Most highly volatile herbicides are no longer used.

Volatility generally increases with increasing temperature and soil moisture, and with decreasing clay and organic matter content (Helling et al. 1971). The use of a surfactant can change the volatility of a herbicide (Que Hee and Sutherland 1981). In extreme cases, losses due to volatilization can be up to 80 or 90% of the total herbicide applied (Taylor and Glotfelty 1988). Of the herbicides described in detail in this handbook, only 2,4-D and triclopyr can present significant volatilization problems in the field (T. Lanini, pers. comm.).

## Behavior in the Environment

Perhaps the most important factor determining the fate of herbicide in the environment is its solubility in water (Hutzinger 1981). Water-soluble

herbicides generally have low adsorption capacities, and are consequently more mobile in the environment and more available for microbial metabolism and other degradation processes. Esters, in general, are relatively insoluble in water, adsorb quickly to soils, penetrate plant tissues readily, and are more volatile than salt and acid formulations (Que Hee and Sutherland 1981).

#### Soils

An herbicide's persistence in soils is often described by its half-life (also known as the DT50). The half-life is the time it takes for half of the herbicide applied to the soil to dissipate. The half-life gives only a rough estimate of the persistence of an herbicide since the half-life of a particular herbicide can vary significantly depending on soil characteristics, weather (especially temperature and soil moisture), and the vegetation at the site. Dissipation rates often change with time (Parker and Doxtader 1983). For example, McCall et al. (1981) found that the rate of dissipation increased until approximately 20% of the applied herbicide remained, and then declines. Nonetheless, half-life values do provide a means of comparing the relative persistence of herbicides.

The distribution of an herbicide in the soil is determined primarily by the amount, type, and surface area of clays and organic matter in the soil, the amount and quality of soil moisture, and soil temperature and soil pH (Helling et al. 1971). Most natural soils have pH values between 5 and 8 (V. Claassen, pers. comm.). Rainfall and the amount of leaching that has occurred strongly influences these values. In wet areas and/or coarse soils, cations can be leached out, leaving the soil acidic. In arid and semi-arid regions, soils retain cations and are more alkaline. Acidic soils can also be found in bogs where organic acids lower the soil's pH.

#### Water

Water bodies can be contaminated by direct overspray, or when herbicides drift, volatilize, leach through soils to groundwater, or are carried in surface or subsurface runoff. Amounts of leaching and runoff are largely dependent on total rainfall the first few days after an application. Total losses to runoff generally do not exceed five to ten percent of the total applied, even following heavy rains (Taylor and Glotfelty 1988). High soil adsorption capacity, low rates of application, and low rainfall reduce total runoff and contamination of local waterways (Bovey et al. 1978).

The behavior of an herbicide in water is dictated by its solubility in water. Salts and acids tend to remain dissolved in water until degraded through photolysis or hydrolysis. Esters will often adsorb to the suspended matter in water, and precipitate to the sediments. Once in the sediments, esters can remain adsorbed to soil particles or be degraded through microbial metabolism. Highly acidic or alkaline waters can chemically alter an herbicide and change its behavior in water. The average pH of surface waters is between five and nine (Hutzinger 1981).

## Environmental Toxicity

The toxicology information reported in this handbook is for the technical grade of the herbicide unless otherwise noted. In some cases, it is not the herbicide itself that is the most toxic component of the applied formula. Adjuvants, such as petroleum solvents (e.g. diesel fuel, deodorized kerosene, methanol), can be highly toxic (Ware 1991). In addition, impurities resulting from the manufacturing process can be more toxic than the active ingredient itself.

#### Birds and Mammals

A herbicide's toxicity is described by its LD50, which is the dose received either orally (taken through the mouth) or dermally (absorbed through the skin) that kills half the population of study animals. The oral LD50s reported here were determined for adult male rats. The dermal LD50s were determined for rabbits. The LD50 is typically reported in grams of herbicide per kilogram of animal body weight. LD50s are determined under varying circumstances so comparisons between different herbicides may provide only a rough sense of their relative toxicities. Dermal LD50 values may be more meaningful to herbicide applicators because they are more likely to be exposed to herbicide through their skin rather than by oral ingestion. In any event, very few people, even among applicators, are exposed to herbicide doses as high as the LD50.

The LD50 does not provide any information about chronic, long-term toxic effects that may result from exposure to lesser doses. Sublethal doses can lead to skin or eye irritation, headache, nausea, and, in more extreme cases, birth defects, genetic disorders, paralysis, cancer, and even death. Impurities derived from the formulation of the herbicide and the adjuvants added to the formulation may be more toxic than the herbicide compound itself, making it difficult to attribute increased risks of cancer or other effects directly to a herbicide (Ibrahim et al. 1991).

The most dramatic effects of herbicides on non-target plants and animals often result from the habitat alterations they cause by killing the targeted weeds. For example, loss of invasive riparian plants can cause changes in water temperature and clarity that can potentially impact the entire aquatic community, and the physical structure of the system through bank erosion. Removing a shrubby understory can make a habitat unsuitable for certain bird species and expose small mammals to predation.

#### Aquatic Species

A herbicide's toxicity to aquatic organisms is quantified with the LC50, which is the concentration of herbicide in water required to kill half of the study animals. The LC50 is typically measured in micrograms of pesticide per liter of water.

In general, ester formulations are more dangerous for aquatic species than salt and acid formulations because ester formulations are lipophilic (fat-loving), and consequently, can pass through the skin and gills of aquatic species relatively easily. Ester formulations, additionally, are not water soluble, and are less likely to be diluted in aquatic systems.

#### Soil Microbes

Herbicides have varying effects on soil microbial populations depending on herbicide concentrations and the microbial species present. Low residue levels can enhance populations while higher levels can cause population declines. In many cases, studies are too short in duration to determine the true long-term impacts of herbicide use on soil microbes.

**HUMAN TOXICOLOGY**

When proper safety precautions are taken, human exposure to herbicides used in natural areas should be minimal. Properly fitted personal protection equipment and well-planned emergency response procedures will minimize exposure from normal use as well as emergency spill situations.

Exposure
Agricultural workers are often exposed to herbicides when they unintentionally re-enter a treated area too soon following treatment. People who mix and apply herbicides are at the greatest risk of exposure. The most common routes of exposure are through the skin (dermal) or by inhalation (to the lungs). Accidental spills or splashing into the eyes is also possible and with some compounds, can result in severe eye damage and even blindness.

Agricultural herbicide applicators are typically exposed to herbicide levels ranging from micrograms to milligrams per cubic meter of air through inhalation, but exposures through the skin are thought to be much greater (Spear 1991). Spilling concentrated herbicide on exposed skin can be the toxic equivalent of working all day in a treated field (Libich et al. 1984). Dermal exposure can occur to the hands (directly or through permeable gloves), splashes onto clothing or exposed skin, and anywhere you wipe your hands (e.g., thighs, brow). Some tests have found relatively high levels of dermal exposure to the crotch and seat of workers who got herbicide on their hands, and then touched or wiped the seat of their vehicles. Because adsorption through the skin is the most common route of exposure for applicators (Marer 1988), the dermal LD50 may provide more practical information on the relative toxicity of an herbicide rather than the oral LD50, which is based on oral ingestion.

Toxic Effects
A person's reaction to pesticide poisoning depends on the toxicity of the pesticide, the size of the dose, duration of exposure, route of absorption, and the efficiency with which the poison is metabolized and excreted by the person's body (Marer 1988, Ware 1991). Different individuals can have different reactions to the same dose of herbicide. Smaller people are, in general, more sensitive to a given dose than are larger people (Marer 1988).

Herbicides can poison the body by blocking biochemical processes or dissolving or disrupting cell membranes. Small doses may produce no response while large doses can cause severe illness or death. The effects may be localized, such as irritation to the eyes, nose, or throat, or generalized, such as occurs when the compound is distributed through the body via the blood stream. Symptoms can occur immediately after exposure or develop gradually. Injuries are usually reversible, but in extreme cases can be permanently debilitating (Marer 1988).

Common symptoms of low-level exposure (such as occurs when mixing or applying herbicides in water) to many herbicides include skin and eye irritation, headache, and nausea. Higher doses (which can occur when handling herbicide concentrates) can cause blurred vision, dizziness, heavy sweating, weakness, stomach pain, vomiting, diarrhea, extreme thirst, and blistered skin, as well as behavioral alterations such as apprehension, restlessness, and anxiety (Marer 1988). Extreme cases may result in convulsions, unconsciousness, paralysis, and death.

Impurities produced during the manufacturing process and adjuvants added to the formulation may be more toxic than the herbicide compound itself. Consequently, LD50s determined for the technical grade of the herbicide may not be the same as that for the brand name formulation. Combinations of herbicides furthermore, can have additive and synergistic effects in which a formulation of two or more herbicides is two to 100 times as toxic as any one of the herbicides alone (Thompson 1996). Labels should be read carefully for manufacturer's warnings and safety precautions that may be required for a particular formulation.

*NOTE: It is important to remember while interpreting study results discussed in this manual and elsewhere that changes in technology have lowered the detectable residue level 1,000-fold over the last twenty years. Herbicide residues that could only be detected to the parts per million (ppm) level (e.g. one microgram of pesticide per kilogram of soil) in the early 1970's can now be detected at the parts per billion (ppb) level (e.g., one microgram of pesticide per 1,000 kilograms of soil). When a study reports finding no residues it really means that no residues above the lowest detectable level were found. This can be an important difference in comparing the results of studies conducted in the 1960's and 70's to studies from the 1980's and 90's.*

## References

Bovey, R. W., C. Richardson, E. Burnett, M. G. Merkle, and R. E. Meyer. 1978. Loss of spray and pelleted picloram in surface runoff water. J. Environ. Qual. 7(2):178-180.
DiTomaso, J. M. 1999. Growth regulators. Presented at the UC Davis Weed Science School, Feb. 24-26 and March 1-3, 1999. Davis, California.
EXTOXNET. 1993. Manifestations of toxic effects. Extension Toxicology Network. Toxicology Information Briefs.
http://ace.orst.edu/info/extoxnet/tibs/manifest.htm
Farmer, W. J., and Y. Aochi. 1987. Chemical conversion of pesticides in the soil-water environment. Chapter 7 in Fate of pesticides in the environment. J. W. Biggar and J. N. Seiber, eds. University of California, Publication 3320.
Freed, V. H., and C. T. Chiou. 1981. Physicochemical factors in routes and rates of human exposure to chemicals. Chapter 3 in Environmental health chemistry. J. D. McKinney, ed. Ann Arbor Science Publishers Inc., Ann Arbor, Michigan.
Helling, C. S., P. C. Kearney, and M. Alexander. 1971. Behavior of pesticides in soil. Adv. Agron. 23:147-240.
Hutzinger, O. 1981. Environmental and toxicological chemistry at the University of Amsterdam: Five years of philosophy and practice of environmental health chemistry. Chapter 2 in Environmental health chemistry. J. D. McKinney, ed. Ann Arbor Science Publishers Inc., Ann Arbor, Michigan.
Kearny, P. C., and J. S. Karns. 1987. Microbial Metabolism. Chapter 10 in Fate of pesticides in the environment. J. W. Biggar and J. N. Seiber, eds. University of California, Publication 3320.
Marer, P. J., M. L. Flint, and M. W. Stimmann. 1988. The safe and effective use of pesticides. University of California, Division of Agriculture and Natural Resources. Publication 3324. 387 pps.
McCall, P. J., S. A. Vrona, and S. S. Kelley. 1981. Fate of uniformly carbon-14 ring labeled 2,4,5-Trichlorophenoxyacetic acid and 2,4-Dichlorophenoxyacetic acid. J. Agric. Food Chem. 29:100-107.
Parker, L. W., and K. G. Doxtader. 1983. Kinetics of the microbial degradation of 2,4-D in soil: effects of temperature and moisture. J. Environ. Qual. 12(4):553-558.
Que Hee, S. S., and R. G. Sutherland. The phenoxyalkanoic herbicides. Vol 1, CRC series in pesticide chemistry. CRC Press, Boca Raton, Fla.
Spear, R. 1991. Recognized and possible exposure to pesticides. Chapter 6 in Handbook of pesticide toxicology, Vol. 1, General Principles. W.J.

Hayes, Jr. and E. R. Laws, Jr. eds. Academic Press, Inc. San Diego, California. 1576 pages.
Taylor, A. W., and D. E. Glotfelty. 1988. Evaporation from soils and crops. Chapter 4 in Environmental chemistry of herbicides, Vol I. R. Grover, ed. CRC Press, Boca Raton, Fla.
Thompson, H. M. 1996. Interactions between pesticides; a review of reported effects and their implications for wildlife risk assessment. Ecotoxicology 5:59-81.
Voos, G. and P. M. Groffman. 1997. Relationships between microbial biomass and dissipation of 2,4-D and dicamba in soil. Biol. Fertil. Soils 24:106-110.
Ware, G. W.. 1991. Fundamentals of pesticides: A self-instruction guide. Thomson Publications. Fresno, California. 307 pgs.
World Health Organization. 1984. 2,4-Dichlorophenoxyacetic acid (2,4-D), Environmental Health Criteria 29. United Nations Environment Programme, Geneva. 151 pgs.
WSSA. 1994. Herbicide Handbook. Weed Science Society of America. Champaign, Illinois. 352pp.

**Date Authored**: April 2001

## The Herbicides

There are approximately a dozen herbicides that TNC has identified in their Weed Control Methods Handbook for use in natural areas. A table of Herbicide Properties for each of these herbicides is provided to give an overview of the characteristics of each of these compounds.

For more specific information on the following for each herbicide;

| | |
|---|---|
| Synopsis | herbicide details (trade names, etc.) |
| History of use | mode of action |
| dissipation mechanisms | behavior in the environment |
| environmental toxicity | application considerations |
| safety measures | human toxicity |

visit http://tncweeds.ucdavis.edu > Invasives and Control Methods > Weed Control Methods Handbook for documents that have been prepared for these specific herbicides.

**Standard Operating Procedures for Herbicide Use**

If you will be using herbicides and want to develop a set of Standard Operating Procedures for Herbicide Use for your site, visit http://tncweeds.ucdavis.edu > Invasives Planning and Strategies > Templates and Examples for a set of procedure developed by TNC's Disney Wilderness Preserve in Florida. These procedures also include a contingency plan in case of spills as well as a generalized check sheet.

**Additional Resources and Information on Management Tools:**

Aquatic Plant Management in Lakes and Reservoirs. Prepared by the North American Lake Management Society and the Aquatic Plant Management Society for the U.S. Environmental Protection Agency, Office of Water Assessment and Watershed Protection Division, 1997. http://aquat1.ifas.ufl.edu/hoyerapm.html.

Biological Control of Invasive Plants in the Eastern United States. 2002. Forest Health and Technology Enterprise Team Publication. FHTET - 2002-04. http://www.invasive.org/eastern/biocontrol/

Center for Invasive Plant Management, http://www.weedcenter.org > Management > Tools > Weed Control Methods (includes additional information on manual, mechanical, grazing, fire, biocontrol, chemical)

Global Invasive Species Program, http://www.cabi-bioscience.ch/wwwgisp/gtcsum.htm > Assessment and Management.

National Invasive Species Center, http://www.invasivespeciesinfo.gov/ > Managers Tool Kit > Control > Control Mechanisms.

Pennsylvania Lake Management Society, http://palakes.org/links.htm.

Southeast Exotic Pest Plant Council, http://www.se-eppc.org/ > Publications > SE-EPPC Invasive Plant Manual > Recommended Herbicide Application Methods.

The Pennsylvania Integrated Pest Management Program, Penn State University and Pennsylvania Department of Agriculture, http://paipm.cas.psu.edu/ > What is IPM?

**Return to top of Page**

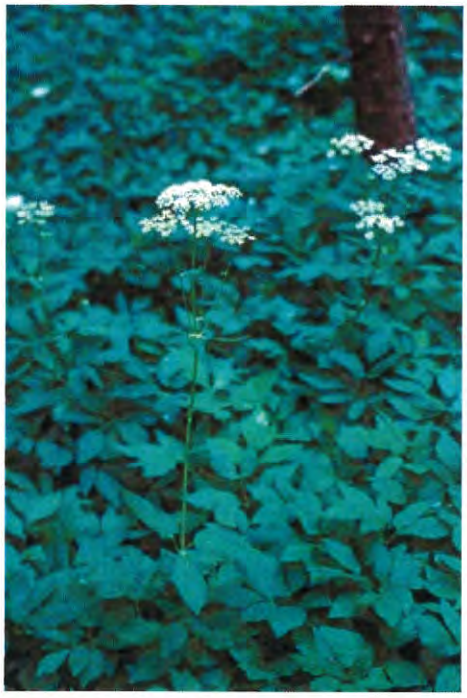
**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Goutweed
## Aegopodium podagraria L.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is April, 2004.

**NATIVE RANGE**: Most of Europe and northern Asia, to eastern Siberia

**DESCRIPTION**: Goutweed, also known as bishop's-weed and snow-on-the-mountain, is an herbaceous perennial plant. It is one of several species of Aegopodium, native to Europe and Asia. Most leaves are basal, with the leafstalk attached to an underground stem, or rhizome. The leaves are divided into three groups of three leaflets, making it "triternate." The leaflets are toothed and sometimes irregularly lobed. Foliage of the "wild" type is medium green in color; a commonly planted variegated form has bluish-green leaves with creamy white edges. Sometimes reversion back to solid green or a mixture of solid green and the lighter variegated pattern occurs within a patch.





Photos: David Schimpf, Department of Biology, University of Minnesota - Duluth, Duluth, MN.

Small, white, five-petaled flowers are produced in mid-summer. Flowers are arranged in flat-topped clusters (called compound umbels) and are held above the ground on a leafy stem up to about 3 feet tall. The seeds are small and elongate, similar in size and shape to carrot seeds, and ripen in late summer. In contrast to the dense foliage cover produced by goutweed, flowering shoots are uncommon in densely shaded areas.



Photo: © John M. Randall/The Nature Conservancy, http://tncweeds.ucdavis.edu/index.html



Photo: Leslie J. Mehrhoff, University of Connecticut, http://invasives.eeb.uconn.edu/ipane/

The rhizomes of goutweed are long, white, and branching, superficially resembling those of quackgrass (Elytrigia repens, also known as Agropyron repens). Patches of goutweed typically form a dense canopy and can exclude most other herbaceous vegetation. Because of this, it is often used as a low-maintenance ground cover.

**ECOLOGICAL THREAT**: Goutweed is an aggressive invasive plant that forms dense patches, displaces native species, and greatly reduces species diversity in the ground layer. Goutweed patches inhibit the establishment of conifers and other native tree species as well.



Photo: © John M. Randall/The Nature Conservancy, http://tncweeds.ucdavis.edu/index.html

**DISTRIBUTION IN THE UNITED STATES**: Goutweed is currently known to occur in twenty-nine states in the mid-Atlantic, Northeast and Northwest (USDA PLANTS map) and is reported to be invasive in natural areas in Connecticut, Michigan, New Jersey, Pennsylvania, Vermont, and Wisconsin (WeedUS Database).

**HABITAT IN THE UNITED STATES**: Goutweed is an ecologically versatile species. It is found in old gardens and flowerbeds, around shrubs and other plantings, and in a variety of other disturbed habitats such as felled forests, abandoned fields, and pastures. In Eurasia, goutweed is primarily a species of deciduous and southern boreal forests, and it expands aggressively in similar habitats in North America. Goutweed appears to do best on moist soil and in light to moderate shade, but is highly shade-tolerant and capable of invading closed-canopy forests.

**BACKGROUND**: Goutweed was apparently first brought to North America as an ornamental during the early stages of European settlement and was well established in the U.S. by 1863. In parts of Russia, the leaves are sometimes used as a salad ingredient and potherb in the spring.

**BIOLOGY & SPREAD**: Goutweed is vigorous, rhizomatous perennial that spreads mainly by vegetative means. Patches increase in size through extension of the rhizome system. The flowers are pollinated by a wide variety of insects, including beetles, bees, and especially small flies. The seeds have no apparent morphological features that would facilitate dispersal. Goutweed seeds require cold stratification to germinate. While established goutweed plants are highly competitive in shaded environments, seedlings generally need recently disturbed soil and rather bright light in order to survive. Goutweed apparently does not form a long-lived seed bank, and the seeds generally germinate the year after ripening. Establishment of goutweed seedlings in the shade is rare. The primary vector for dispersal to new areas is humans. Most goutweed colonies spread to neighboring natural areas from intentional plantings, or by the dumping of yard waste that includes discarded rhizomes.

**AUTHORS**: Steve Garske, Great Lakes Indian Fish & Wildlife Commission, Odanah, WI. David Schimpf, Department of Biology, University of Minnesota - Duluth, Duluth, MN.

**REVIEWER**: Wayne Owen, U.S.D.A. Forest Service, Washington, DC.

**EDITOR**: Jil M. Swearingen, National Park Service, National Capital Region, Center for Urban Ecology, Washington, DC

**For more information on garlic mustard, please contact**:
David Schimpf, Department of Biology, University of Minnesota - Duluth, Duluth, MN (218-726- 7265; dschimpf@d.umn.edu)
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Steve Garske, Great Lakes Indian Fish & Wildlife Commission, Odanah, WI (715-682-6619; steveg@glifwc.org)

**REFERENCES:**
Bailey, W.W. 1987. Journal of the Torrey Botanical Society. Vol. 3: 176. Borg-Karlson, A. K., I. Valternova, and L. A. Nilsson. 1994. Volatile compounds of flowers of six species in the family Apiaceae: bouquets for different pollinators? Phytochemistry (Oxford) 35: 111-118.
Dlussky, G. M. 1998. Mechanisms of competition for pollinators in Anthriscus sylvestris Hoffm. and Aegopodium podagraria L. (Apiaceae). Zhurnal Obshchei Biologii 59: 24-44. Everett, T. H. 1981. The New York Botanical Garden illustrated encyclopedia of horticulture. Vol. 1. A-Be. Garland Publishing Inc., New York.
Gatsuk, L. E., O. V. Smirnova, L. I. Vorontzova, L. B. Zaugolnova, and L. A. Zhukova. 1980. Age states of plants of various growth forms: a review. Journal of Ecology 68: 675-696.
Gleason, H. A., and A. Cronquist. 1991. Manual of vascular plants of northeastern United States and adjacent Canada, 2nd ed. New York Botanical Garden, New York.
Grime, J. P., J. G. Hodges, and R. Hunt. 1988. Comparative plant ecology. Unwin Hyman, London.
Jansson, O. 1974. Phylloquinone (vitamin K1) levels in leaves of plant species differing in susceptibility to 2,4-dichlorophenoxyacetic acid. Physiologia Plantarum 31: 323-325. Meyer, K., and F. H. Hellwig. 1997. Annual cycle of starch content in rhizomes of the forest geophytes Anemone nemorosa and Aegopodium podagraria. Flora (Jena) 192: 335-339.
Shishkin, B. K. (ed.). 1950. Flora of the USSR. Volume XVI-Umbelliflorae. Botanical Institute of the Academy of Sciences of the USSR. (Translated from Russian by Israel Program for Scientific Translations, Jerusalem, 1973.)
Small, E. 1973. Photosynthetic ecology of normal and variegated Aegopodium podagraria. Canadian Journal of Botany 51: 1589-1592.
Swearingen, J. 2004. WeedUS: Database of Invasive Plants Affecting Natural Areas in the U.S. (In progress) http://www.nps.gov/plants/alien
Tutin, T. G., V. H. Heywood, N. A. Burges, D. M. Moore, D. H. Valentine, S. M. Walker, and D. A. Webb (eds.) 1968. Flora Europaea, Vol. 2. Cambridge University Press, Cambridge.

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Goutweed
# *Aegopodium podagraria* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resources:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Goutweed - http://www.nps.gov/plants/alien/fact/aepo1.htm - Authors: Steve Garske and David Schimpf - April 1, 2004.**

## GENERAL MANAGEMENT CONSIDERATIONS:

A variety of methods are available for controlling goutweed, depending on the extent of the infestation and the amount of time and labor available. Regardless of the control method used, the patch should be carefully monitored periodically for a few years. New shoots should be dug up and destroyed. Once goutweed control has been achieved, revegetation with native or non-invasive exotic plant materials is recommended. This is particularly important on sites where erosion is a concern or where other invasive species are likely to colonize the site if left alone.

## MANUAL AND MECHANICAL CONTROL:

Manual. Small patches of goutweed can be eliminated by careful and persistent hand-pulling or digging up of entire plants along with underground stems (rhizomes). Pulled plants can be piled up and allowed to dry for a few days before bagging and disposing of them. Be careful to pick up all rhizomes which, if left behind, can reroot and sprout new plants. For large patches, a team of volunteers or use of herbicide is recommended.

Mechanical. Where appropriate, frequent short mowing may control or slow the spread of goutweed in lawns, along roadsides, and other areas.

Physical. Preventing goutweed from photosynthesizing in early spring (at the time of leaf-out) can control the plant by depleting its carbohydrate reserves. This can be accomplished by covering the patch with black plastic sheeting when the leaves start to emerge from the ground in the spring, and leaving it in place through the summer. A more effective option is to cut all plants once they've fully leafed out, using a mower, scythe, or weed-whacker type machine, and then cover the area with plastic. Covering the plants in mid- or late summer, after they have regained substantial starch reserves, is probably much less effective.

## BIOCONTROL:

There are no biological control organisms currently available for Aegopodium podograria in North America.

## CHEMICAL CONTROL:

Systemic herbicides such as glyphosate (Roundup) that are translocated to the roots and kill the entire plant are most effective for goutweed control. However, glyphosate is non-specific and can damage or kill desirable native plants that are accidentally sprayed in the course of treating the goutweed. Contact herbicides are usually ineffective because goutweed readily leafs out again after defoliation.

Return to List

# DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers

## Garlic Mustard

**Alliaria petiolata (M. Bieb.) Cavara & Grande**

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is April, 2004.

**NATIVE RANGE:** Europe

**DESCRIPTION:**
Garlic mustard is a cool season biennial herb in the mustard family (Brassicaceae) with stalked, triangular to heart-shaped, coarsely toothed leaves that give off an odor of garlic when crushed.



Photo: Dan Tenaglia, www.missouriplants.com, www.invasives.org




Photo: Chris Evans, Univ. of Georgia, www.invasives.org




Photo: Chris Evans, The Univ. of Georgia, www.invasives.org

First-year plants appear as a rosette of green leaves close to the ground. Rosettes remain green through the winter and develop into mature flowering plants the following spring.




Photo: Jody Schimp, Illinois Dept. of Natural Resources, www.invasive.org

Flowering plants of garlic mustard reach from 2 to 3-1/2 feet in height and produce buttonlike clusters of small white flowers, each with four petals in the shape of a cross.




Photo: Chris Evans, The Univ. of Georgia, www.invasives.org



Photo: Britt Slattery,
U.S. Fish and Wildlife Service, www.invasive.org

Beginning in May (in the mid-Atlantic Coast Plain region), seeds are produced in erect, slender pods and become shiny black when mature. By late June, when most garlic mustard plants have died, they can be recognized only by the erect stalks of dry, pale brown seedpods that remain, and may hold viable seed, through the summer.

## Recognition of garlic mustard is critical. Several white-flowered native plants, including toothworts (Dentaria), sweet cicely (Osmorhiza claytonii), and early saxifrage (Saxifraga virginica), occur alongside garlic mustard and may be mistaken for it.

**ECOLOGICAL THREAT**: Garlic mustard poses a severe threat to native plants and animals in forest communities in much of the eastern and midwestern U.S. Many native widflowers that complete their life cycles in the springtime (e.g., spring beauty, wild ginger, bloodroot, Dutchman's breeches, hepatica, toothworts, and trilliums) occur in the same habitat as garlic mustard. Once introduced to an area, garlic mustard outcompetes native plants by aggressively monopolizing light, moisture, nutrients, soil and space. Wildlife species that depend on these early plants for their foliage, pollen, nectar, fruits, seeds and roots, are deprived of these essential food sources when garlic mustard replaces them. Humans are also deprived of the vibrant display of beautiful spring wildflowers.




Photo: Victoria Nuzzo, Natural Area Consultants, www.invasive.org.

Garlic mustard also poses a threat to one of our rare native insects, the West Virginia white butterfly (Pieris virginiensis). Several species of spring wildflowers known as "toothworts" (Dentaria), also in the mustard family, are the primary food source for the caterpillar stage of this butterfly. Invasions of garlic mustard are causing local extirpations of the toothworts, and chemicals in garlic mustard appear to be toxic to the eggs of the butterfly, as evidenced by their failure to hatch when laid on garlic mustard plants.

**DISTRIBUTION IN THE UNITED STATES**: Garlic mustard ranges from eastern Canada, south to Georgia and as far west as Oregon. Click here to see another distribution map.

**HABITAT IN THE UNITED STATES**: Garlic mustard frequently occurs in moist, shaded soil of river floodplains, forests, roadsides, edges of woods and trails edges and forest openings. Disturbed areas are most susceptible to rapid invasion and dominance. Though invasive under a wide range of light and soil conditions, garlic mustard is associated with calcareous soils and does not tolerate high acidity. Growing season inundation may limit invasion of garlic mustard to some extent.

**BACKGROUND**: Garlic mustard was first recorded in the United States about 1868, from Long Island, New York. It was likely introduced by settlers for food or medicinal purposes.

**BIOLOGY & SPREAD**: After spending the first half of its two-year life cycle as a rosette of leaves, garlic mustard plants develop rapidly the following spring into mature plants that flower, produce seed and die by late June. In the mid-Atlantic Coastal Plain region, seeds are produced in erect, slender, four-sided pods, called siliques, beginning in May. Siliques become tan and papery as they mature and contain shiny black seeds in a row. By late June, most of the leaves have faded away and garlic mustard plants can be recognized only by the dead and dying stalks of dry, pale brown seedpods that may remain and hold viable seed throughout the summer. A single plant can produce thousands of seeds, which scatter as much as several meters from the parent plant. Depending upon conditions, garlic mustard flowers either self-fertilize or are cross-pollinated by a variety of insects. Self-fertilized seed is genetically identical to the parent plant, enhancing its ability to colonize an area. Although water may transport seeds of garlic mustard, they do not float well and are probably not carried far by wind. Long distance dispersal is most likely aided by human activities and wildlife. Additionally, because white-tailed deer prefer native plants to garlic mustard, large deer populations may help to expand it by removing competing native plants and exposing the soil and seedbed through trampling.

**AUTHORS**: Pamela Rowe, Montgomery County Department of Environmental Protection, Rockville, MD. Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on garlic mustard, please contact:**
Cornell University, Biological Control of Weeds--Garlic Mustard, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Southeast Exotic Pest Plant Council; Invasive Plant Manual, http://www.se-eppc.org/manual/
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Nonnative Invasive Plants of Southern Forests, http://www.invasive.org/eastern/srs/
Victoria Nuzzo, Cornell University, Ithaca, NY; vnuzzo@earthlink.net
Virginia Natural Heritage Program Fact Sheet--Garlic Mustard; http://www.dcr.virginia.gov/dnh/invlist.htm

**REFERENCES**:
Abugatta, Alonso. 1996. Progress made in educating about problems with invasive plants. Virginia Native Plant Society Newsletter, Vol. 15, No. 3, (May) p. 5.
Byers, D.L. 1987. The effect of habitat variation in Alliaria petiolata on life history characteristics. American Journal of Botany 74:647.
Nuzzo, V.A. 1991. Experimental Control of garlic mustard in Northern Illinois using fire, herbicide, and cutting. Natural Areas Journal 11(3):158-167.
Nuzzo, V.A. 1993. Distribution and spread of the invasive biennial garlic mustard (Alliaria petiolata) in North America, pp. 137-146. In B.N. McKnight (ed.), Biological Pollution. Indiana Acad. Sci. Indianapolis, Indiana, 261 pp.
Nuzzo, V.A. 1996. Impact of dormant season herbicide treatment on the alien herb garlic mustard (Alliaria petiolata) [Bieb] Cavara & Grande. Transactions of the Illinois State Academy of Science, vol 89, pp. 25-36.
Tennessee Exotic Pest Plant Council. 1997. Tennessee Exotic Plant Management Manual, pp. 56-59.
The Nature Conservancy. Garlic Mustard: Element Stewardship Abstract. In: Wildland Weeds Management & Research Program, Weeds on the

Recommended control procedures:




## Species Management and Control Information

### Garlic Mustard
### *Alliaria petiolata* (M. Bieb.) Cavara & Grande

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

Alliaria petiolata should be removed from natural areas before it sets seed, as one plant can populate or repopulate a site. It appears to threaten the structure of forest communities, and should be given high management priority as soon as it is observed in or near a preserve.

The goal of Alliaria petiolata management is to prevent seed production. The primary management objective in areas lacking this plant is to prevent establishment, by annually monitoring for and removing all Alliaria petiolata plants. The primary management objective in infested sites is to prevent seed production. Cutting flowerstalks is effective in small populations. Fire and herbicide may be useful for larger populations but both have potential side effects. No method provides 100% control.

Growing season mortality reduces Alliaria petiolata seedling populations by 80%-90% between spring and late fall (Anderson et al. 1996, Byers and Quinn 1998); hence, control is most economical when undertaken in late fall or early spring prior to flower production. Late fall is usually the preferred season for control, as native plants are dormant and management can be conducted until snow covers the ground. If weather is unfavorable in fall, control can still be conducted in early spring. Delaying control until spring can be risky, as native herbs may begin growth earlier than anticipated, and weather may limit or prevent management activities.

Management priority should be given to small populations of Alliaria petiolata, and to high quality natural areas. Low quality natural areas, and sites with established large Alliaria petiolata populations, have low management priority.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weed Gone Wild - Fact sheet - Garlic mustard- http://www.nps.gov/plants/alien/fact/alpe1.htm - Authors: Pamela Rowe and Jil M. Swearingen - April 1, 2004.**

Because the seeds of garlic can remain viable in the soil for five years or more, effective management requires a long term commitment. The goal is to prevent seed production until the stored seed is exhausted. Hand removal of plants is possible for light infestations and when desirable native species co-occur. Care must be taken to remove the plant with its entire root system because new plants can sprout from root fragments. This is best achieved when the soil is moist, by grasping low and firmly on the plant and tugging gently until the main root loosens from the soil and the entire plant pulls out. Pulled plants should be removed from site if at all possible, especially if flowers are present.

For larger infestations of garlic mustard, or when hand-pulling is not practical, flowering stems can be cut at ground level or within several inches of the ground, to prevent seed production. If stems are cut too high, the plant may produce additional flowers at leaf axils. Once seedpods are present, but before the seeds have matured or scattered, the stalks can be clipped, bagged and removed from the site to help prevent continued buildup of seed stores. This can be done through much of the summer.

**Southeast - Exotic Pest Plant Council - Invasive Plant Manual - Garlic mustard - http://www.se-eppc.org/manual/garlicmustard.html - 2003**

Hand Pull: This method of control is effective for small populations of garlic mustard, since plants pull up easily in most forested habitats. Hand-pulling is an extremely effective method of reducing population and seed productivity. Hand-pulling of plants can be done during most of the year. If plants have seed capsules present, they should be bagged and disposed of to prevent seed dispersal. Care should be taken to minimize soil disturbance. Resprouts are uncommon but may appear from mature plants not entirely removed.

Cutting: This method is effective for medium- to large- sized populations depending on available time and labor resources. Cut stems when in flower (late spring/early summer) at ground level either manually or with a mechanical brush-cutter. This technique will result in almost total mortality of existing plants. Dormant seeds in the soil are unaffected by this technique. Treatments should be continued annually until the seedbank is exhausted.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

**CUTTING**

Cutting flowering Alliaria petiolata plants at ground level results in 99% mortality, and eliminates seed production. Cutting at 10 cm above ground level results in 71% mortality and reduces seed production by 98% (Nuzzo 1991). Cutting is most effective when plants are in full bloom and/or have developed siliques; plants cut earlier in the flowering period may have sufficient resources to produce additional flowerstems from buds on the root crown (Nuzzo, personal observation).

Cut flower stems may form viable seed. Solis (1998) pulled Alliaria petiolata plants at four flowering stages (flowerbud, flowering, newly formed siliques, well-formed siliques) and piled each flowering stage in a separate fenced plot; the following spring Alliaria petiolata seedlings were abundant in all plots and absent from the empty control plot, indicating that pulled (and by implication, cut) Alliaria petiolata flower stems produce viable seed. Cavers et al. (1979) suggested that vivipary (germination of seeds while still in the silique) does not occur, although all seeds remained viable during the observation period. Until more information is available, cut or pulled stems should be removed from the site whenever feasible.

Cutting with a weed whip provides quick removal of flowering stems, but may remove other desirable species. Some native species, such as Trillium, are severely impacted if cut. Most other species are not substantially damaged, and the benefits of removing Alliaria petiolata should be weighed against the temporary reduction in growth and reproduction of native groundcover species. Cutting with a weed whip may distribute cut stems (and hence seeds) across the site; care should be taken to collect and remove cut material whenever possible.

**MANIPULATION OF WATER LEVEL AND SALINITY**

Not tested. Short-term growing season inundation does not harm Alliaria petiolata rosettes (Byers and Quinn 1998). Extended mid summer flooding resulted in 100% mortality of immature rosettes, but also distributed Alliaria petiolata seeds across the floodplain (Nuzzo 1999).

**MOWING, DISCING AND PULLING**

Mowing may have an effect comparable to cutting, but the equipment could severely disturb soil and native vegetation. Mowing would be useful in full sun situations such as roadsides, but may distribute seeds. In forested locations a weed whip is more practical and less damaging. Discing is not an acceptable method of control due to the destruction of the community. Root cutting is a very effective although labor intensive means of control.

Pulling is very labor intensive but effective if the upper half of the root is removed. Alliaria petiolata frequently snaps off at or just below the root crown when the flower stalk is pulled, leaving adventitious buds which send up new flower stalks. Pulling can result in substantial soil disturbance, damaging desirable species and bringing up Alliaria petiolata seeds from the seedbank. Soil should be thoroughly tamped after pulling to minimize chances for re establishment of garlic mustard or other weedy species. In general, cutting is a less destructive method of control than pulling but is effective only when the flower stalk is elongating, whereas pulling can be conducted throughout the growing season.

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/gmustard.html - Vol.1, No. 6 - Authors: Victoria Nuzzo, Jill Kennay, George Fell - December 20, 1990.**

Research by Victoria Nuzzo indicates that cutting flowering stems at ground level results in 99% mortality, while cutting at 4 inches (10 cm) above ground level produces 71% mortality and reduces total seed production by 98%. Plants cut near ground level when in full flower usually do not resprout. Viable seed may be produced after stems are cut: pending further research, cut stems should be removed from the site when possible. The Nature Conservancy has successfully controlled or eliminated this plant from several sites by a combination of spring burning, hand-pulling, and cutting flowering stems with a scythe. When garlic mustard occurs in nearly pure populations with few other plants, scything is advantageous in that large areas can be covered quickly and the soil is not disturbed.

Initial effort in areas of light infestation:
Removal of plants by hand-pulling is effective if the root is removed. If the stem snaps off from the root crown of a non-flowering plant, the plant may resprout. When hand-pulling, disturb the soil as little as possible, and tamp the soil firmly after removing the plant. Soil disturbance can bring garlic mustard seed to the surface and create a favorable environment for garlic mustard germination and growth.

## PRESCRIBED FIRE:

**Plant Conservation Alliance, Alien Plant Working Group - Weed Gone Wild - Fact sheet - Garlic mustard-**

**http://www.nps.gov/plants/alien/fact/alpe1.htm - Authors: Pamela Rowe and Jil M. Swearingen - April 1, 2004.**

Fire has been used to control garlic mustard in some large natural settings but, because burning opens the understory, it can encourage germination of stored seeds and promote growth of emerging garlic mustard seedlings. For this reason, burns must be conducted for three to five consecutive years. Regardless of the control method employed, annual monitoring is necessary for a period of at least five years to ensure that seed stores of garlic mustard have been exhausted.

**Southeast - Exotic Pest Plant Council - Invasive Plant Manual - Garlic mustard - http://www.se-eppc.org/manual/garlicmustard.html - 2003**

In fire-tolerant communities, prescribed burning can be effective either alone or in conjunction with herbicide. Mid-intensity spring burns appear to reduce density of adult plants somewhat more effectively than fall burns. A program of repeated seasonal burning over several years is most effective in deterring garlic mustard and enhancing growth of native ground-layer vegetation.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

Prescribed burning provides inconsistent management results; Alliaria petiolata populations may remain stable, decrease, or increase following one or more fires. In Kentucky, Alliaria petiolata populations had no significant change following three years of repeated dormant season burns (Luken and Shea 2000). In Illinois, Alliaria petiolata populations were maintained at a low level when fires burned completely through the affected area, and were conducted for at least two consecutive years (Nuzzo 1991, Nuzzo et al. 1996, Schwartz and Heim 1996). Fires that left a thin litter layer (1-2 cm) did not kill Alliaria petiolata root crowns, which subsequently produced multiple flower stalks from axillary buds (Nuzzo et al. 1996). Fires that removed the litter layer may enhance survival of seedlings that germinate after burning (Nuzzo et al. 1996). Thus, after a single fire, total Alliaria petiolata cover can increase due to survival of adult plants, and/or to enhanced seedling survival.

Spring and fall fires are equally successful in reducing cover of Alliaria petiolata rosettes (Nuzzo 1991, Nuzzo et al. 1996). Spring fires also reduce seedling presence if conducted during the germination period (Nuzzo 1991), although native herbaceous species are damaged by a growing season burn (Schwartz and Heim 1996).

Use of fire as a management tool should be tailored to the specific community. Removal of the litter layer may facilitate invasion by disturbance adapted species, including Alliaria petiolata, particularly if there is little native ground layer present at the site. Fires should only be conducted when at least two or more consecutive fires can be scheduled; burning only once may increase Alliaria petiolata abundance. Impact of consecutive fires on the community should be considered, including changes in ground layer composition. While fire can maintain Alliaria petiolata at low abundance, it does not eliminate this plant (Nuzzo et al. 1996).

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/gmustard.html - Vol.1, No. 6 - Authors: Victoria Nuzzo, Jill Kennay, George Fell - December 20, 1990.**

Initial effort in areas of heavy infestation
Fall or early spring burning is an effective control treatment in oak woods. Repeated burns over several years may be necessary to achieve adequate control and to eliminate plants produced from the seed bank. Prescribed fires should be of sufficient intensity to burn the affected site thoroughly. Low intensity fires that leave unburned areas will not control garlic mustard effectively. Any isolated plants that are not burned should be removed by hand prior to flower production.

## BIOCONTROL:

**Van Driesche, R., et al., 2002, Biological Control of Invasive Plants in the Eastern United States, USDA Forest Service Publication FHTET-2002-04, 413 p. http://www.invasive.org/eastern/biocontrol/29GarlicMustard.html.**

No releases of agents have yet been made against garlic mustard

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

Biological weed control - the use of a plant's specialized "natural enemies" to control a nonnative plant - may be the only effective way to reduce large infestations Alliaria petiolata in North America. B. Blossey of Cornell University is coordinating a program to investigate the potential for biological control of Alliaria. At least 69 phytophagous insect species and seven fungi are associated with Alliaria petiolata in Europe (Hinz and Gerber 1998). Of these, four monophagous weevils (two shoot-miners, one seed-feeder, and one root feeder; all Ceutorhynchus species) are being investigated as potential biocontrols. Ceutorhynchus alliariae and C. robertii are sibling species that mine shoots and petioles of rosettes and bolting plants of garlic mustard; heavily attacked plants die without producing seeds (Hinz and Gerber 1998). C. constrictus larvae develop in Alliaria petiolata seeds; each larva destroys one-two seeds (Hinz and Gerber 1998). C. scrobicollis larvae feed in the root crown and occasionally the lower parts of shoots; heavily attacked plants die without producing seeds (Hinz and Gerber 1999). Adults of all 4 Ceutorhynchus species feed on Alliaria petiolata leaves and cause some damage, but the majority of damage is caused by the larvae (Hinz and Gerber 1999). A fifth species, the flea beetle Phyllotreta ochripes, preferentially feeds on Alliaria petiolata, but adults have been found on other Cruciferae, including Rorippa, Brassica, and Sinapsis species (Hinz and Gerber 1999). All five insects will be studied for several years in Europe to document their impact on Alliaria petiolata, and to verify that Alliaria petiolata is their exclusive food source. If one or more insects pass all safety and specificity tests required by the USDA, they will be brought to North America for field releases.

An unidentified virus (or several viruses) can kill a flowering plant and prevent it from ripening viable seeds in Ontario (Cavers personal communication 1989), and may provide some control. Alliaria petiolata developed severe mosaic symptoms when infected with CMV and TuMV, but only when grown in ruderal soil; plants in a natural environment had mostly symptomless infection (Polak 1985).

## CHEMICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weed Gone Wild - Fact sheet - Garlic mustard- http://www.nps.gov/plants/alien/fact/alpe1.htm - Authors: Pamela Rowe and Jil M. Swearingen - April 1, 2004.**

For very heavy infestations, where the risk to desirable plant species is minimal, application of the systemic herbicide glyphosate (e.g., Roundup) is also effective. Herbicide may be applied at any time of year, including winter (to kill overwintering rosettes), as long as the temperature is above 50 degrees F. and rain is not expected for about 8 hours. Extreme care must be taken not to get glyphosate on desirable plants as the product is non-selective and will kill almost any plant it contacts. Spray shields may be used to better direct herbicide and limit non-intentional drift.

**Southeast - Exotic Pest Plant Council - Invasive Plant Manual - Garlic mustard - http://www.se-eppc.org/manual/garlicmustard.html - 2003**

Glyphosate Foliar Spray: This method is effective on populations where mechanical control measures are not feasible or are impractical. Apply a 2% solution of glyphosate and water plus a non-ionic surfactant using a tank or backpack sprayer to thoroughly cover all leaves. Do not apply so heavily that herbicide drips off the leaf surface. Glyphosate is a non-selective herbicide requiring caution not to spray non-target species. Treatments should be done in the early spring when most other non-target vegetation is dormant. Refer to manufacturer's label for specific information and restrictions regarding use.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

Dormant season herbicide application can provide effective control of Alliaria petiolata, but poses a potential threat to native herbaceous and graminoid species. Glyphosate (Roundup, Rodeo, Accord) produces a high degree of control, but results in some native herb loss and significantly reduces graminoid cover. Bentazon (Basagran SG, Basagran T/O) produces nearly equal control, with much lower impact on graminoid species, and little or no impact on native herbs. Glyphosate is suitable for use in forest communities that have few semi evergreen herbaceous or graminoid species. Bentazon appears suitable for use in many forest communities, but should be subjected to further tests before widespread use. The effectiveness of chemical control is dependent on the chemical contacting all plants; under field conditions this rarely or never happens.

Glyphosate (Roundup, Rodeo, Accord)

Glyphosate (Roundup) applied at 1%, 2%, and 3% concentrations to dormant rosettes in late fall or early spring reduced adult cover by >95% (Nuzzo 1991, 1996). Control was slightly greater with higher glyphosate concentrations. Seedlings that germinate after application are not affected by the herbicide because glyphosate is inactivated when it contacts soil. Glyphosate applied after germination will significantly reduce seedling populations (Nuzzo 1991).

Glyphosate is a non-selective herbicide and will kill native species, particularly when applied in spring. At the community level, glyphosate did not affect mean species richness or total mean herbaceous cover, but did significantly reduce cover of both sedges and grasses, at both 0.5% and 1% concentrations (Nuzzo 1996).

2,4 D (sold under many brand names)

2,4 D is not recommended for control of Alliaria petiolata. An ester formulation of 2,4 D applied during the growing season reduced Alliaria petiolata cover by 10% at 0.25 lb/acre, 70% at 0.50 lb /acre, and 45% at 1.0 lb /acre. Dormant season application at very low concentration had no effect on Alliaria petiolata (Nuzzo unpublished). Ortho Weed B Gone (a 2,4 D formulation available at many hardware and garden stores) at 0.4% had no effect on Alliaria petiolata adult plants (Rich Dunbar personal communication 1990).

Mixing 2,4 D with other chemicals provides a more effective kill, based on visual assessment. A 1% solution of Mecamine (2,4 D plus Dicamba) reduced Alliaria petiolata (Bill McClain personal communication 1990). Treatment with Kilmore (2,4 D, MCPP and Dicamba) applied at the rate of 1.1 liter/hectare killed all flowering Alliaria petiolata (R.H. Brown personal communication in Cavers et al. 1979).

Triclopyr (Garlon 3A, Garlon4, Pathfinder)

A spring application of triclopyr amine (Garlon 3A ) mixed at 7oz/5gallon water (just over 1%) killed 92% of Alliaria petiolata rosettes in a limited test (Rich Dunbar personal communication 1990).

Bentazon (Basagran SG and Basagran T/O)

Growing season application of Bentazon (Basagran) at 0.50 1.0 lb /acre reduced Alliaria petiolata rosette cover by 90 95% (Nuzzo 1994). Impacts of dormant season application could not be determined by this study, as severe drought killed rosettes in both control and treatment plots (Nuzzo 1996). Bentazon did not affect species richness or herb cover, and had minimal effect on graminoid cover (Nuzzo 1996). Alliaria petiolata seedlings were not affected by treatment (Nuzzo 1996). Bentazon is a post emergent contact herbicide that kills dicots (broadleaf plants) and some sedges but will not kill most grasses. It is used to control mustards in agricultural fields and is primarily a contact herbicide, meaning it kills or injures portions of the plant that it lands on and little else. It blocks photosynthesis by binding with the D1 protein of the photosystem II complex. This stops CO2 fixation and production of ATP and NADPH2 which are needed for plant maintenance and growth. In most cases, however, the plants are actually killed because the blockage in the photosystem promotes formation of oxidized molecules that cause chain reactions destroying chlorophyll, carotenoids and cellular membranes. Bentazon's biggest drawback appears to be that it is very soluble in water and does not bind to soil particles which suggests a high potential for groundwater contamination. It has a short half life and breaks down rapidly in soil (average half-life 20 days), however, and estimates based on an EPA survey indicate it is found in about 0.1% of rural drinking water wells

nationwide although it is widely used. It was not detected in any US community water systems nor was it detected at concentrations above 0.02 mg/L in any well. Bentazon is rapidly broken down in non-susceptible plants and has little effect on germinating seeds. The brands Basagran SG and Basagran T/O are registered for use in non-crops sites, roadways and other rights of way and thus can be legally used in natural areas in at least some states. The labels state that this herbicide should not be applied under sycamores or rhododendrons and that it may kill trees and shrubs if applied to soil beneath their canopies.

Acifluorfen (Blazer)

Acifluorfen is not recommended for control of Alliaria petiolata. Like Bentazon, Aciflourfen is used to control mustards in agricultural fields. Warm season application of Acifluorfen (Blazer) at 0.25, 0.37 and 0.5 lb /acre reduced Alliaria petiolata cover by 30 40% (Nuzzo 1994). Dormant season treatment eliminated Alliaria petiolata rosettes, and prevented seedling germination the next spring (no seedlings were observed 7 months after fall treatment, and 2 months after spring treatment) (Nuzzo 1996). Aciflourfen reduced species richness and graminoid cover, and significantly reduced herbaceous cover in treated plots (Nuzzo 1996). Aciflourfen has a soil residual, and treated plots had low herbaceous cover through the first growing season after treatment (Nuzzo 1996).

Mecoprop (Mecomec, many other brand names)

Alliaria petiolata was heavily damaged by aerial application of 2.4 kg /acre of mecoprop (Birnie 1984).

**Nonnative Invasive Plants of Southern Forests: a field guide for identification and control: Gen. Tech. Rep. SRS-62. Asheville, NC, U.S. Department of Agriculture, Forest Service Research Station - http://www.invasive.org/eastern/srs/GM.html - Author: James H. Miller - 2003.**

Recommended control procedures:

- To control two generations, thoroughly wet all leaves with a glyphosate herbicide as a 2-percent solution in water (8 ounces per 3-gallon mix) during flowering (April through June). Include a surfactant unless plants are near surface waters.
- In locations where herbicides cannot be used, pull plants before seed formation. Repeated annual prescribed burns in fall or early spring will control this plant, while "flaming" individual plants with propane torches has also shown preliminary success.

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/gmustard.html - Vol.1, No. 6 - Authors: Victoria Nuzzo, Jill Kennay, George Fell - December 20, 1990.**

Spot application of 2% Roundup (a formulation of glyphosate) to the foliage of individual plants is effective during spring and fall when most native vegetation is dormant but garlic mustard remains green. Herbicide should be applied when air temperatures are above 32o F (0o C). Managers should exercise caution when applying herbicide to garlic mustard to avoid contacting nontarget plants. Roundup is a nonselective herbicide (kills all vegetation) and should not be used during the growing season in high-quality areas because of the possibility of harming nontarget plants. Do not spray so heavily that herbicide drips off the target species. The herbicide should be applied while backing away from the treated areas to avoid contacting the wet herbicide. By law, herbicides only may be applied as per label instructions and by licensed herbicide applicators or operators when working on public properties.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Weed Alert: Garlic mustard (Alliaria petiolata) - http://tncweeds.ucdavis.edu/alert/alrtalli.html - Tunyalee Martin - April 2000 .**

Control:
Preventing infestations by monitoring and removing pioneering plants is the best control. Where populations are established, seed production must be prevented for 2-5 years to deplete the seed bank.

1)Minimize disturbance by reducing foot traffic, overgrazing and erosion.
2)Monitor once or twice annually for seedlings and new invasions. Check for garlic mustard in very early spring or late fall for immature rosettes. Monitor in early to mid spring for flowering adults. Immediately remove small populations before seeds are produced.
3)Hand pulling works well but must be continued until the seed bank is exhausted. The plant pulls easiest early in the season. Remove the plant and at least the upper half of the root. Adventitious buds on the upper half of the root can send up new flower stalks if not removed.
4)Natural mortality of seedlings is high (approximately 95%). Chemical control is most economical in late fall or early spring.
5)Fair control was observed with a 3% concentration of glyphosate (Roundup) applied with a sprayer in Indiana. A decline in population was observed with a 5% solution of glyphosate (Roundup) sprayed on in Kentucky. Glyphosate is a non-selective herbicide.
6)2,4-D is not effective for the control of garlic mustard.
7)A spring application of triclopyr (Garlon 3A) killed 92% of garlic mustard rosettes in a small test. Triclopyr was applied at 7 oz/5 gal water (a little over 1%). 8)Cutting before seed set is effective if the infestation is minor. Cutting of large infestations are not as effective. Repeated cuttings at ground level produce the best control results. If feasible, cut stems should be removed.
9)Burning can provide control if it burns completely through the affected area. Scattered plants may survive in some areas. In Wisconsin, plants have been observed resprouting after a burn. New seedlings may germinate after a fire.

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/gmustard.html - Vol.1, No. 6 - Authors: Victoria Nuzzo, Jill Kennay, George Fell - December 20, 1990.**

Maintenance control
Vigilant monitoring and hand removal of first- and second-year plants prior to flower production can be effective. A regular burning regime in oak woods can control garlic mustard.

Recommended Practices on Buffer and Severely Disturbed Sites
Fall or early spring burning in oak woods can control this species. Repeated burns may be necessary over several years. Spot application of 2% Roundup to individual plants as described above can be used in severely disturbed woods. Cutting or scything flowering stems, as described above, is effective. Maintenance control is the same as given above. In addition, hand spraying individual plants with an amine formulation of 2,4-D is an effective control when applied according to label instructions. To reduce vapor drift, use an amine formulation of 2,4-D rather than an ester formulation. A 1% solution of Mecamine (2,4-D plus Dicamba) applied to the foliage of young plants is also effective. Either herbicide should be applied only during spring or fall when most native vegetation is dormant but garlic mustard remains green. The herbicide 2,4-D amine is selective for broadleaf plants. As with Roundup, managers should exercise caution when applying these herbicides to garlic mustard to avoid contacting nontarget plants. Do not spray so heavily that herbicide drips off the target species.

Failed or Ineffective Practices
Low intensity fires that do not burn through the leaf litter have no effect on garlic mustard.

Return to List




## Cheatgrass (Drooping brome-grass)
## *Bromus tectorum* L.

**SOURCE**: The following information on this species is taken from the Invasive Plant Atlas of New England (IPANE) website at http://invasives.eeb.uconn.edu/ipane/. Modifications include additional pictures (www.invasive.org) and resource information. Check the website links for future updates.

**DESCRIPTION**: *Bromus tectorum* is an erect stemmed annual grass that can grow from 20-70 cm (8-25 in.) in height. Its leaf sheaths and blades are covered with soft short hairs. The leaves can be from 2-4 mm (0.08-0.16 in.) wide and up to 20 dm (8 in.) long. Its ligules are from 1 to 2.5 mm (0.04-0.1 in.) long. The panicles measure 5-20 cm (2-7.75 in.) long, have numerous branches, but retain an open quality and are generally nodding. The panicles bear from 3 to 8 drooping spikelets, each spikelet is from 2 to 3.5 cm (0.8-1.4 in.) long. The glumes are awl-shaped; the lemmas are toothed, narrowly lanceolate and hairy. The lemmas 1-1.5 mm (0.04-0.06 in.) wide, acuminate and lead to slender, straight awns that are 10-17 mm (0.4-0.67 in.) long.

*Bromus tectorum* flowers from May to June. It's foliage and inflorescences are a shiny light green up to the flowering period, but then turn a wine-red color after flowering, and eventually turn brown as they senesce. Most of the plants usually die and fall over by July. The seeds of *Bromus tectorum* can germinate in the fall or in the spring; fall germination is generally more common. Its fibrous root system is finely divided. When a seed germinates in the fall, the developing root system is able to expand over the winter, giving the plant an increased ability to exploit available water and nutrients in the spring.

Page References Fernald 103, Gleason & Cronquist 773, Holmgren 723, Magee & Ahles 163. See reference section below for full citations.



Photos: Leslie J. Mehrhoff, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/

**SIMILAR SPECIES:** *Bromus* spp

**DISTRIBUTION:** *Bromus tectorum* is a native of Mediterranean Europe. It gets the name "tectorum", which means roof, from its historical penchant for growing on thatched roofs. It is currently present in all fifty of the United States, all of the Canadian provinces and parts of Mexico. It is much more abundant in the western U.S. and Canada, notably in regions where the annual rainfall is between 15 and 55 cm (5.9-21.7 in.). It is most problematic on heavily grazed rangeland and regions where winter wheat is grown. *Bromus tectorum* is on the noxious weed lists of at least 35 states. In New England, it is found mostly along roadsides and in disturbed areas.

**HISTORY OF INTRODUCTION IN NEW ENGLAND:** *Bromus tectorum* was first introduced to North America sometime before 1860; in the years that followed, it was probably introduced many times, at a wide range of different sites. There is evidence that *Bromus tectorum* arrived as a seed lot contaminant, and also in ballast soil. It may have been helped to spread around the country by trains, because it was often found in straw that was used in the rail system as packing material. The earliest North American records come from wheat field regions in Washington, Utah, Colorado, Montana and British Columbia. The common name "cheatgrass" comes from western farmers who thought they had been given

impure seed when *Bromus tectorum* started spreading into their wheat fields. It spread quickly during the late 19th and early 20th century. By 1920 to 1930, *Bromus tectorum* had, arguably, become the worst weedy range grass in the western United States. It is currently the dominant species on more than 100 million acres of land in the intermountain West.

The first records of *Bromus tectorum* are from New York and Pennsylvania in 1861. There are many other records of *Bromus tectorum* in the Northeast from the 1870s, mostly in sandy, waste sites not far from the coast. Robinson and Fernald (1908) described it as being found in "waste places from Maine to Illinois and southward". By 1950, Fernald described it as being found at "roadsides and waste places, often too abundant". It is presently common in all of the states of New England, though it does not pose the sorts of problems in the Northeast that it does out West.

**HABITATS (IN NEW ENGLAND):** Abandoned Field, Agricultural Field, Edge, Open Disturbed Area, Pasture, Railroad Right-of-Way, Utility Right-of-Way, Vacant Lot, Yard or Garden

Within New England, *Bromus tectorum* is mostly found in disturbed sites, waste areas and along roadsides. It does well in areas with dry, sandy soil.

**THREATS:** *Bromus tectorum* has the ability to draw down soil moisture and nutrients to very low levels, making it difficult for other species to compete. Due to its tendency to mature early and then dry out, it gains a competitive advantage through the promotion of fire. An increased cycle of fires favors annual species at the expense of many perennials. Though very important to *Bromus tectorum's* success out West, this fire dynamic has not helped *Bromus tectorum* in New England. *Bromus tectorum* is a serious agricultural weed in the western U.S., but in New England it is starting to have an impact in nursery and orchard settings.



Photos: Leslie J. Mehrhoff, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/

**For more information on cheatgrass, contact:**
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
Montana State Weed Science, http://weeds.montana.edu/crop/brome.htm
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Bugwood Network, MA-EPPC Invasive Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Northeastern Area, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Tech, Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm

**REFERENCES:**
Fernald, M.L. 1950. Gray's Manual of Botany 8th edition. American Book Company, New York.
Gleason, H. A. 1952. The New Britton and Brown Illustrated Flora of the Northeastern United States and Adjacent Canada. Macmillan Publishing Co., Inc. New York
Gleason H.A. and A.C. Cronquist. 1991. Manual of Vascular Plants of the Northeastern United States and Adjacent Canada. 2nd ed. New York Botanical Garden, Bronx, New York. 773.
Holmgren N.H. 1998. Illustrated Companion to Gleason and Cronquist_s Manual. New York Botanical Garden, Bronx, New York.
Hulbert, L. 1955. Ecological studies of Bromus tectorum and other annual bromegrasses. Ecol. Monogr. 25:181-213.
Mack, R. N. 1981. Invasion of Bromus tectorum L. into western North America: an ecological chronicle. Agro-Ecosyst. 7:145-165.
Magee D.W and H.E. Ahles. 1999. Flora of the Northeast. University of Massachusetts Press, Amherst.
Morrow, L. A. and P W. Stahlman. 1984. The history and distribution of downy brome (Bromus tectorum) in North America. Weed Sci. 32:2-6.
Robinson, B.J. 1908. Gray's New Manual of Botany, 7th ed. American Book Co., New York, NY.
USDA, NRCS. 2001. The PLANTS Database, Version 3.1. (http://plants.usda.gov). National Plant Data Center, Baton Rouge, LA 70874-4490




## Species Management and Control Information

# Cheatgrass (Drooping brome-grass)
# *Bromus tectorum* L.

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Montana State University - Weed Science - Bromus tectorum - http://weeds.montana.edu/crop/brome.htm - Authors: Meghan Trainor, Alvin J. Bussan - September 19, 2001**

Prevention

| Despite successful efforts to control downy brome in croplands -- infested rangelands, pastures, roadsides, and waste areas throughout Montana provide a continuing source of downy brome seed for reinvasion of winter wheat crops. Seed spread is mainly through contaminated grain, hay, straw, manure and farm machinery. The long awn attached to the seed helps it stick to animal fur, as well.

To prevent initial infestation in cultivated land, ensure that any seed purchased is certified free of downy brome and thoroughly clean equipment, especially swathers and combines. Four main reasons are cited as to why downy brome has become more of a major problem in cultivated land.

1. Farmers are re-cropping more, thereby not allowing for the opportunity to break up the downy brome life cycle.
2. The shorter wheat varieties used today allow more light to penetrate the crop canopy, making downy brome more competitive.
3. More fertilizer is used each year giving downy brome a competitive edge.
4. Farmers rarely use the mold-board plow and are instead using variations of either stubble/mulch tillage or no tillage at all. The chisel plow doesn't invert soil, and as a result, downy brome seed is no longer buried deeply. In no-till farming, downy brome seed remains on the soil surface which is the preferred environment of this plant since it is a surface germinator. It thrives under no-till production. Shallow burial, or surface germination, is the ideal germination environment for downy brome (Wright and Fay 1995).

Cultural Control

Cropland Weed Specialist, A.J. Bussan at MSU in Bozeman, says the first key to successful control of downy brome it to select appropriate fields. Crop rotation is the principal management strategy for downy brome. For example, downy brome populations increased from 2 up to 60 plants per ft2 after 3 years of continuous winter wheat in research trials. In contrast, winter wheat - fallow or winter wheat - canola rotations maintained downy brome populations below 10 plants ft2 in the same study. As a result, a number of Montana growers have adopted a 3 or 4 year rotation that alternates spring crops with winter wheat and fallow. In selecting fields for planting winter wheat, growers should select fields where the downy brome has been effectively cleaned up by crop rotation.

Another approach involves timely summer fallow tillage. The major cause of the downy brome problem is annual seed production. Timely summer fallow tillage will break this cycle. Downy brome matures quickly in the spring (hence the weed's other common name Junegrass). Therefore, summer fallow tillage must occur early enough to prevent seed production. If seed heads emerge from a downy brome plant before tillage or after tillage, viable seed will be produced and must be dealt with in the fall prior to seeding winter wheat. Pre-plant control is another effective strategy. The most common technique used by Montana farmers to control downy brome is to delay winter wheat seeding in the fall until rains occur. This stimulates germination of downy brome seed which is easily controlled during winter wheat seedbed preparation. Bussan suggests preparing a firm seedbed 2 weeks prior to planting. The tillage and mixing of wet and dry soil can stimulate downy brome seed to germinate and emerge. The emerged downy brome can be managed by additional tillage or herbicide application prior to planting. Popular herbicides for pre-plant management include Roundup or Gramoxone Extra. Both products can be applied after planting, but before crop emergence, as well. Roundup and Gramoxone Extra should be applied with ammonium sulfate. Be sure to follow the label for proper rates.

For growers with limited or no-till cropping systems, stale seed bed techniques may not fit. However, using a Phoenix or spring-toothed harrow prior to planting may stimulate emergence, in particular if rain occurs prior to planting. In addition, growers could use stale seedbed techniques on field edges or in problem areas where downy brome is likely to occur to prevent its spread across fields.

Narrow row spacing of winter wheat reduces the competitive ability of downy brome. However, this is not a feasible practice in the dryer areas of Montana due to increased competition among wheat plants for moisture.

For those farmers who anticipate a downy brome problem, increasing the seeding rate of winter wheat can be an effective technique. Increasing the seeding rate of winter wheat creates more competition for the downy brome plants.

Downy brome evolved in a rangeland situation and is therefore adapted to that type of environment. Thus, the seed germinates easily on the soil surface, but less effectively when buried. Researchers in Washington report that firmer seedbeds allowed fewer downy brome seedlings to emerge.

Since downy brome grows along roadsides, it serves as a source of seed which can infest adjacent cultivated land. The summer fallowing pattern of cultivated land can eliminate much of the downy brome seed that's produced along the borders of fields.

## MANUAL AND MECHANICAL CONTROL:

**Montana State University - Weed Science - Bromus tectorum - http://weeds.montana.edu/crop/brome.htm - Authors: Meghan Trainor, Alvin J. Bussan - September 19, 2001**

Neither mowing nor burning are effective controls to prevent seed set. Seed will continue to mature on cut-off plants. One option is mowing and removal of plants from the field. Burning is not effective, as many of the seeds drop to the ground before the heat can kill them.

As discussed above early spring tillage of fallow ground, tillage prior to planting spring-season crops, and tillage operations that bury downy brome seeds (mold-board plowing) are effective mechanical methods.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Bromus tectorum - http://tncweeds.ucdavis.edu/esadocs/bromtect.html - Authors: Alan T. Carpenter and Thomas A. Murray - 1999.**

CONTROL WITH CUTTING: Cutting is not a recommended control method for cheatgrass. Plants that are cut before seed ripening will regenerate new culms and produce seeds at the cut height. Plants that are cut after seed ripening will die, but by this point the seeds are already viable. For more information, see Lawrence Memorial Grassland Experiment in the Management Programs Section.

CONTROL WITH GRAZING, DREDGING, AND DRAINING: *Bromus tectorum* is considered to be valuable forage in some ranching operations (Emmerich et al. 1993). However, grazing is not a recommended method of control for cheatgrass. If the plants are grazed in the spring, they will regenerate new culms and produce seeds. When grazed in the summer or fall the plants will not regrow, but by then viable seeds have already been produced. Also, the long awns of the seeds on the mature plants may damage the mouths and intestinal tracts of the livestock.

There are no references to indicate that dredging or draining have been tested, or would be an adequate control method for *Bromus tectorum*. Cheatgrass does not usually grow in sites that could be dredged or drained.

CONTROL WITH MANIPULATION OF WATER LEVEL AND SALINITY: There are no references to indicate this has been tested, or would be an adequate control method for *Bromus tectorum*.

CONTROL WITH MOWING, DISKING, AND PULLING: Mowing is not usually an effective control of *Bromus tectorum* (Whitson et al. 1997). Although cheatgrass plants will die if they are mowed after ripening, by then the seeds are already viable. When mowed at an earlier growth stage, the plants can regenerate new culms and produce viable seeds. Therefore, mowing once a year does not prevent stands from producing viable seeds (Whitson et al. 1997).

In one study, repeated mowing (every three weeks) during the spring and summer was as effective at controlling cheatgrass seed production as an application of glyphosate (Ponzetti 1997). However, this method was very labor-intensive and a cost/benefit analysis should be conducted before any choice is made.

Hand-pulling cheatgrass plants in small infestations before seed set would effectively eliminate current seed production, but may not eliminate the infestation. The large seed bank commonly associated with cheatgrass infestations will allow plants to re-establish for several years without noticeable reductions in plant density. Hence, any pulling program must be conducted for several years, or until the seed bank has been exhausted. Also, seeds that blow into the cleared areas from adjacent uncleared areas may negate the effects of pulling. When pulling, an effort should be made to extract as much of the root as possible so that the plant can not simply regrow and produce new seeds.

## PRESCRIBED FIRE:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Bromus tectorum - http://tncweeds.ucdavis.edu/esadocs/bromtect.html - Authors: Alan T. Carpenter and Thomas A. Murray - 1999.**

Bromus tectorum is a highly flammable species due to its complete summer drying, its fine structure, and its tendency to accumulate litter (5). A fire will reduce the plants to ash, but fire intensity may not be great enough to consume the litter layer, and seeds in the soil will probably survive (5). The amount of litter or ash left on a site is a good indicator of the amount of cheatgrass seed still surviving (5).

Wendtland (1993) studied the effects of spring, summer, and fall burns on mixed-grass prairie in western Nebraska that had been tilled in the distant past and had subsequently reverted to prairie ("go-back" lands). The study sites had abundant cheatgrass. He found that both summer and fall burns were effective at controlling cheatgrass without severely damaging most native species plants. However, he recommended fall burns because they were much easier to control than summer burns. Interestingly, blue grama (*Bouteloua gracilis*), which is often the dominant plant over vast areas of short-grass prairie, was damaged significantly more by burning than other native plant species.

In Colorado, burning is usually conducted in June after the plant has dried, but before the seeds have dropped (Beck pers. comm.). However, some seeds will survive and if a burn is not followed by reseeding cheatgrass will recover to pretreatment proportions within 3 to 4 years (Beck pers. comm.). Reseeding should be done in late fall (a dormant seeding) (Beck pers. comm.).

Cheatgrass fires may reduce the cover of valuable perennial species e.g., shrubs like sagebrush and grasses like bluebunch wheatgrass, that are not adapted to frequent fires (5). Additionally, areas that have been burned become susceptible to erosion until they green up again.

Cheatgrass fires can burn very rapidly and can be very dangerous. Firefighters have reported cheatgrass fires that traveled between 20-40 mph, over-running firefighters and equipment (Devine 1998). Controlled cheatgrass burns should always be conducted by trained individuals.

## BIOCONTROL:

**Montana State University - Weed Science - Bromus tectorum - http://weeds.montana.edu/crop/brome.htm - Authors: Meghan Trainor, Alvin J. Bussan - September 19, 2001**

The studies on potential biological controls of downy brome have mainly focused on downy brome in a rangeland environment. One such study looked at potential biological control of downy brome with crown and root rot fungi. The researchers suggested that crown rot might be a potential biological control on downy brome in the arid western U.S. because it is adapted to dry soils and has low virulence on desirable grasses, such as wheatgrass (Grey et al. 1995). Thus, crown rot causes greatest damage on moisture-stressed plants and results in susceptible disease reactions on downy brome.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Bromus tectorum - http://tncweeds.ucdavis.edu/esadocs/bromtect.html - Authors: Alan T. Carpenter and Thomas A. Murray - 1999.**

No biological control agents are available for use against cheatgrass at present. The biological control of weeds is based on the premise that insect feeding stresses or kills plants or reduces seed output and eventually causes a reduction in weed density (Berube and Myers 1982). Biological agents have never been known to completely eliminate the host species, but can significantly reduce their abundance. In fact, complete elimination would be self-defeating to the control agent. However, if different stresses on plants are cumulative, biological controls which lower the competitive ability of weeds should enhance the effectiveness of other control methods.

In laboratory and small-scale field tests, phytotoxins produced by the naturally occurring rhizobacteria, *Pseudomonas fluorescens* (strain D7), and *Pseudomonas syringae* (strain 3366) adversely affected cheatgrass at several growth stages (Gealy et al. 1996, Gealy et al. 1995). These bacteria produce plant suppressive compounds (PSC's) that selectively inhibit the germination and early root growth of cheatgrass, and thus shift the competitive advantage back to perennial grasses (Ogg et al. 1991). The active compound produced by these bacteria appears to be a phenazine-1-carboxylic acid (Gealy et al. 1996). When purified from strain 3366 and applied at a rate of 5.7 mg/L, the acid inhibited downy brome root growth by 99% (Gealy et al. 1996).

The phytotoxins produced by strain D7 and strain 3366 inhibit lipid synthesis and disrupt membrane integrity (Gealy et al. 1996). These two effects can stop cheatgrass seedling growth within several hours after exposure (Gealy et al. 1996). Further tests indicated that cheatgrass root growth is about 200 times more sensitive to D7 than shoot growth (Gealy et al. 1996). Gealy et al. (1996) determined that older seedlings were inhibited several orders of magnitude less than very young seedlings. The greatest suppression of cheatgrass occurred in cool (10/10 or 18/13°C) (50/50 or 64.4/55.4°F) or moist conditions (Johnson et al. 1993). Since cheatgrass seeds usually germinate and establish in the fall, the application of bacterium would need to be timed to coordinate with seedling emergence.

Neither of these two rhizobacteria are currently approved as biological controls for cheatgrass. The focus of the research on them has been to develop a biological control for cheatgrass in winter wheat crops and the effects of these rhizobacteria on desirable plants in noncrop applications has not been determined. Additional research may be able to determine if these agents can provide effective control of cheatgrass on the ground in noncrop situations and natural areas.

## CHEMICAL CONTROL*:

**Montana State University - Weed Science - Bromus tectorum - http://weeds.montana.edu/crop/brome.htm - Authors: Meghan Trainor, Alvin J. Bussan - September 19, 2001**

Herbicide programs have been developed for downy brome control in alfalfa, perennial grasses grown for seeds, and for some rangeland plants. In winter wheat, however, chemical control is difficult due to the species having similar growth habits and susceptibility to herbicides.

The herbicides that control downy brome include atrazine, bromacil, cyanazine, chloropropham, diclofop, glyphosate, metribuzin, paraquat (with or without selected triazines), pronamide, propham, simazine, terbacil, and trifluralin (Wicks 1984; Pepper 1984; Swan and Whitesides 1988).

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Bromus tectorum - http://tncweeds.ucdavis.edu/esadocs/bromtect.html - Authors: Alan T. Carpenter and Thomas A. Murray - 1999.**

There are several types of herbicides that can be used alone or combined to provide effective control of *Bromus tectorum*. For relatively small

infestations, a backpack sprayer is recommended to minimize the danger to non-target plants. However, infestations are often so large that a four-wheeler, tractor, or truck fitted with a sprayer is necessary. The following herbicides are divided into two groups, spring applied and fall applied.

Spring Applied Herbicides: In most cases, applications should be made in early spring when non-target species are dormant to ensure selective control. Cheatgrass was reported to be controlled best when the plants were 10 cm (3.9 in) high or less and growing vigorously at the time of application (Wiese et al. 1995).

Quizalofop: Trade Name: Assure II®: Whitson et al. (1988) reported that quizalofop applied in mid-April at a rate of 0.5 lb./acre controlled 100% of the cheatgrass in the study. However, the herbicide suppressed seed head production of perennial grasses (Whitson et al. 1988). In another study, quizalofop at 0.02 lb./acre provided greater than 95% control of cheatgrass. No mention was made of damage to perennial grasses (Wiese et al. 1995).

Quizalofop is a grass meristem destroyer, and is usually used to remove grass species from any non-grass crop (Ross and Childs 1996). All grass meristem destroyers should be used post-emergence on seedling grasses before the seed head is detectable in the top leaf sheath (boot stage) (Ross and Childs 1996). Once applied, leaves yellow, redden, and sometimes wilt (Ross and Childs 1996). Quazalofop does not damage most broadleaf species and is registered in the United States for noncrop use outside California (DuPont, 1999a).

Fluazifop-p-butyl: Trade names: Fusilade® 2000, Fusilade® DX: Fluazifop-p-butyl is another postemergence herbicide that attacks the meristematic tissue of annual and perennial grass species (Ross and Childs 1996). Fluazifop-p-butyl is actively taken up and translocated throughout the plant (7). It accumulates in the actively growing regions and interferes with the plant cell's ability to produce energy (7). Fluazifop-p-butyl does not kill broadleaved plants, and at sublethal rates will suppress seed head development in cheatgrass (Ahrens 1994). At very low rates, fluazifop-p-butyl retards grass growth (Ahrens 1994). However, if considering using low rates of fluazifop-p-butyl against cheatgrass, the herbicide's effects on desireable non-target grass species should be determined on a site by site basis.

The application rate in noncrop situations is generally 16-24 oz/acre (1-1.5 lb./acre). Like quizalofop, fluazifop-p-butyl should be applied early post-emergence, before the seed head is detectable in the top leaf sheath (Ross and Childs 1996). Fluazifop-p-butyl is not registered for use on *Bromus tectorum* in California.

Sethoxydim: Trade names: Poast®, Poast Plus®: Sethoxydim is a postemergence herbicide used to control annual and perennial grass species (8). Like quizalofop and fluazifop it does not damage most broadleaved species (forbs and woody plants). Sethoxydim is commonly used to control grass weeds in broad-leaved vegetable, fruit, field and ornamental crops (8). Sethoxydim has been used in noncrop situations as well. It is registered for "set aside conservation reserve land" in the Midwest, South and Northeast. Except in California, it is registered throughout the U.S. for noncrop use (BASF, 1999).

Sethoxydim shows some selectivity among grass species, particularly among cool season grasses (Ross and Childs 1996). Sethoxydim should be applied early postemergence before the grass reaches the boot stage.

Paraquat: Trade Name: Gramaxone®: Paraquat applied at 0.5 to 0.7 lb./acre controlled greater than 97% of cheatgrass when applied during late April or early May (Whitson et al. 1993). Blackshaw (1991) reported that paraquat at 0.22 to 0.27 lb./acre controlled cheatgrass by 80-90% when applied up to the 3-5 tiller stage.

Paraquat is a contact herbicide that kills only the tissue contacted. Paraquat penetrates into the cytoplasm and causes the formation of peroxides and free electrons which destroy the cell membranes almost immediately (Ross and Childs 1996). Severe injury is evident hours after application and maximum kill is attained within a week (Ross and Childs 1996). If the plant is only partially covered with herbicide, only partial shoot kill will occur. Any new growth on the surviving plants will be normal in appearance, and foliar applications alone can only provide shoot kill (Ross and Childs 1996).

Glyphosate: Trade Names: Roundup®, Roundup Ultra®, Rodeo®, Accord®: Glyphosate applied when cheatgrass plants have 3-5 tillers at a rate of 0.16 to 0.18 lb./acre controlled cheatgrass by 80-90% (Blackshaw 1991). However a different experiment by Beck et al. (1995) using glyphosate at a much higher rate of 0.37 to 0.5 lb./acre resulted in only 80% reductions of cheatgrass within and between years. Glyphosate is a non-selective herbicide and will damage or kill desirable vegetation that it contacts including forbs and woody species. It should be applied in early spring after cheatgrass has established, but before perennial seedlings have emerged.

Glyphosate is an amino acid inhibitor (Ross and Childs 1996). It is a relatively non-selective compound that is used to control annual grasses and broad-leaved plants. Uses are limited to foliar applications only, as it is quickly inactivated in the soil (Ross and Childs 1996). Symptoms include yellowing of new growth and death within days to weeks (Ross and Childs 1996).

Imazameth: Trade Name: Plateau®: Imazameth can be applied at a rate of 0.24 to 0.75 lb./acre to newly established or existing stands of big bluestem (*Andropogon gerardii*), little bluestem (*Schizachyrium scoparium*), Indiangrass (*Sorghastrum nutans*), sideoats grama (*Bouteloua curtipendula*), and blue grama (*Bouteloua gracilis*) to control cheatgrass, as well as other annual and perennial weeds (1,6). Plateau® is new herbicide that provides a broad spectrum of weed control for roadsides and native grasses in noncrop applications (3). It controls annual and perennial weeds without adverse effects on cool or warm season perennial grasses (1,2). Brian Winter, a TNC steward in Minnesota, used Plateau® to control leafy spurge (Euphorbia esula). He notes that it effectively controls cool season grasses and forbs, but leaves the warm season native grasses. It is labeled for use during the re-establishment of native grass prairie, but is not labeled for use in pastures, rangeland, or along streamsides (2).

Imazameth is an amino acid inhibitor that has a long residual life in the soil, and may leach into groundwater. It should not be used around streams and rivers (2). When applied, shoot meristems cease growth and roots tend to develop poorly (Ross and Childs 1996). Complete symptom development can be very slow and may take two to three weeks to develop (Ross and Childs 1996).

Fall Applied Herbicides: Fall herbicide applications should be conducted after cheatgrass seeds have germinated and are beginning to grow. Fall

applications are generally used in cropland situations by farmers growing winter wheat or other cool season crops. However, sometimes these herbicides are used in pastures and rangelands.

Sulfometuron Methyl: Trade Name: Oust®: Sulfometuron methyl is a pre-emergence and post-emergence herbicide that controls many annual and perennial grasses and broadleaf weeds in noncrop areas (Dupont 1999b). Apply sulfometuron methyl at a rate of 3 to 5 oz/acre (0.2-0.3 lb./acre) for cheatgrass control (9). Masters (1998) found sulfometuron methyl has only a minimal effect on native perennial grasses and is helpful, when fall applied with imazapyr, in re-establishing native grasses.

Sulfometuron methyl is an extremely potent herbicide and can damage non-target vegetation if it is not applied correctly. Users should read the product label carefully before applying sulfometuron methyl to any area.

Atrazine: Trade Name: Aatrex®: In one experiment, atrazine at a rate of 0.54 lb./acre was the most cost-effective herbicide for decreasing competition of annual brome grasses and increasing yields of perennial grasses (Currie et al. 1987). In addition to suppressing cheatgrass, atrazine seems to stimulate protein production on native shortgrass range (Currie et al. 1987).

Atrazine is a pre-emergence, and to a limited extent early post-emergence, photosynthetic inhibitor that is mainly used in crops, but is sometimes used in pastures, rangeland, and noncropland (Ross and Childs 1996). It is registered for use on roadsides in Colorado, Kansas, Montana, North Dakota, Nebraska, South Dakota and Wyoming. In conservation reserve programs, Atrazine may be used in Nebraska, Oklahoma, Oregon and Texas (Novartis, 1999). Atrazine kills cheatgrass seedlings in the fall after they emerge from the soil while perennial plants are dormant. The following spring, the area is free of cheatgrass, and clear for the establishment of native perennial grasses. The half-life of atrazine in the soil ranges from 60-100 days. Atrazine is highly mobile in soil and has a high potential for groundwater contamination.

More information on chemical control of cheatgrass can be obtained from the Weed Management Library at 1-800-554-WEED.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




## Spotted knapweed
## Centaurea maculosa Larmarck or C. biebersteinii DC.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is May 2003.

**NATIVE RANGE**: Central Europe, east to central Russia, Caucasia, and western Siberia
**DESCRIPTION**: Spotted knapweed is a biennial or short-lived perennial. Its name is derived from the spots formed by black margins on the flower bract tips.




Photo: Cindy Roche, www.forestryimages.org




Photo: Jim Story, Montana State University, Cindy Roche, www.forestryimages.org

Spotted knapweed typically forms a basal rosette of leaves in its first year and flowers in subsequent years. Rosette leaves are approximately 8 inches long by 2 inches wide, borne on short stalks, and deeply lobed once or twice on both sides of the center vein, with lobes oblong and wider toward the tip. The taproot is stout and deep.



Photo: Linda Wilson, University of Idaho, www.forestryimages.org.




Photo: Great Smoky Mountains National Park, Resource Management Archives, National Park Services, www.forestryimages.org.

Flowering stems are erect, 8 to 50 inches tall, branched above the middle, and sparsely to densely hairy. Stem leaves alternate along the stem, are unstalked, and may be slightly lobed, or linear and unlobed. Leaf size decreases towards the tip of the stem.

Flowers are purple to pink, rarely white, with 25 to 35 flowers per head. Plants bloom from June to October, and flower heads usually remain on the plant. Flower heads are oblong or oval shaped, ¼ inch wide and ½ inch across, and are single or borne in clusters of two or three at the branch ends. Leaf like bracts surrounding the base of the flower head are oval and yellow green, becoming brown near the base. The margins of these bracts have a soft spine like fringe, with the center spine being shorter than the lateral spines. The brown, oval seeds are 1/16 to 1/8 inch long, with pale longitudinal lines and a short fringe on one end.

**ECOLOGICAL THREAT**: Spotted knapweed infests a variety of natural and semi-natural habitats including barrens, fields, forests, prairies, meadows, pastures, and rangelands. It outcompetes native plant species, reduces native plant and animal biodiversity, and decreases forage production for livestock and wildlife. Spotted knapweed may degrade soil and water resources by increasing erosion, surface runoff, and stream sedimentation. It has increased at an estimated rate of 27% per year since 1920 and has the potential to invade about half of all the rangeland (35 million acres) in Montana alone.




Photo: Norman E. Rees, USDA - ARS, www.forestryimages.org.

**DISTRIBUTION IN THE UNITED STATES**: Spotted knapweed is a widely distributed species reported to occur throughout Canada and in every state in the U.S. except Alaska, Georgia, Mississippi, Oklahoma and Texas (see map). Click here to see a distribution map. It has been designated as a noxious weed in Arizona, California, Colorado, Idaho, Minnesota, Montana, Nebraska, Nevada, New Mexico, North Dakota, Oregon, South Dakota, Utah, Washington, and Wyoming.
It has been identified as invasive in natural areas by eighteen organizations in twenty-six states (Arizona, California, Colorado, Connecticut, Delaware, Kentucky, Idaho, Illinois, Massachusetts, Maryland, Michigan, Minnesota, Montana, North Carolina, New Jersey, New York, Oregon, Pennsylvania, South Dakota, Tennessee, Utah, Virginia, Washington, Wisconsin, West Virginia, and Wyoming). Fifteen national parks also identify spotted knapweed as an invasive plant and a threat to natural habitats.

**HABITAT IN THE UNITED STATES**: Spotted knapweed is found at elevations up to and over 10,000 feet and in precipitation zones receiving 8 to 80 inches of rain annually. Spotted knapweed prefers well-drained, light-textured soils that receive summer rainfall, including open forests dominated by ponderosa pine and Douglas fir, and prairie habitats dominated by Idaho fescue, bluebunch wheatgrass, and needle-and-thread grass. Disturbance allows for rapid establishment and spread; however, spotted knapweed is capable of invading well managed rangelands. Spotted knapweed does not compete well with vigorously growing grass in moist areas. In seasonally dry areas, spotted knapweed's taproot allows it to access water from deep in the soil, beyond the reach of more shallowly rooted species.

**BACKGROUND**: Spotted knapweed was introduced to North America from Eurasia as a contaminant in alfalfa and possibly clover seed, and

through discarded soil used as ship ballast. It was first recorded in Victoria, British Columbia in 1883 and spread further in domestic alfalfa seeds and hay before it was recognized as a serious problem.

**BIOLOGY & SPREAD**: Spotted knapweed plants in North America generally live 3 to 7 years but can live up to nine years or longer. Plants regrow from buds on the root crown. Reproduction is by seed, and plants are capable of producing 500- 4,000 seeds per square foot per year. About 90% of the seeds are viable at the time of dispersal, and they can remain viable in the soil for 5-8 years. Most seeds are dispersed near the parent plant but can be transported by people, wildlife, livestock, vehicles, and in soil, crop seed, and contaminated hay. Gravel pits, soil stockpiles, powerlines, grain elevators, railroad and equipment yards are important seed distribution points.

**AUTHOR(S):**
Michael Carpinelli, USDA-ARS, Burns, OR (541-573-8911, michael.carpinelli@oregonstate.edu).

**EDITOR**:
Jil M. Swearingen, National Park Service, National Capital Region, Natural Resources and Science, Center for Urban Ecology, Washington, DC.

**REVIEWERS:**
Ruth Douglas, Charlottesville, VA.
Karen Enyedy, Austin, TX.
Jim Jacobs, Montana State University, Bozeman, MT.
. Lori Makarick, National Park Service, Grand Canyon National Park, Grand Canyon, AZ.
Phillip Moore, AHTD, AR.
Monica Pokorny, Montana State University, Bozeman, MT.
Leila M. Shultz, Harvard University Herbaria, Cambridge, MA.
Ed Shadrick, V3 Consultants, Woodridge, IL.

**For more information on garlic mustard, please contact:**
Cornell University, Biological Control of Weeds--Garlic Mustard, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Delaware River Invasive Plant Partnership - http://www.paflora.org/DRIPP.html
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Michael Carpinelli, USDA-ARS, Burns, OR (541-573-8911, michael.carpinelli@oregonstate.edu)
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Peter Rice, Montana Noxious Weed Trust (http://invader.dbs.umt.edu/)
Steve Dewey's Weed Web (http://www.ext.usu.edu/ag/weeds/index.htm)
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
Virginia Natural Heritage Program Fact Sheet - Spotted knapweed - http://www.dcr.virginia.gov/dnh/invlist.htm
Virginia Tech Weed Identification Guide - http://www.ppws.vt.edu/scott/weed_id/cenma.htm
Wisconsin Department of Natural Resources - Non-native Plant Fact Sheets - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm

**REFERENCES**:
Boggs, K.W. and J.M. Story. 1987. The population age structure of spotted knapweed (Centaurea maculosa) in Montana. Weed Sci. 35:194 198.
Chicoine, T.K. 1984. Spotted knapweed (Centaurea maculosa L.): Control, seed longevity and migration in Montana. MS Thesis Montana State University. Bozeman, Mont. 96 pp MIN 25 8430038.
Chicoine, T.K., P.K. Fay, and G.A. Nielsen. 1985. Predicting weed migration from soil and climate maps. Weed Sci. 34:57 61.
Groh, H. 1944. Canadian weed survey. 2nd Ann. Rep. Can. Dep. Agric.
Kartesz, J.T. 1994. A Synonymized Checklist of the Vascular Flora of the United States, Canada, and Greenland. Vol.1 - Checklist. 2nd ed. Timber Press, Portland, OR. 622 p.
Lacey, J.R., C.B. Marlow, and J.R. Lane. 1989. Influence of spotted knapweed (Centaurea maculosa) on surface water runoff and sediment yield. Weed Technol. 3:627 631.
Lacey, C.A., J.R. Lacey, P.K. Fay, J.M. Story, and D.L. Zamora. 1995. Controlling knapweed in Montana rangeland. Circular 311, Cooperative Extension Serv., Montana State Univ. Bozeman. Mauer, T., M. J. Russo, and M.Evans. 1987. Element Stewardship Abstract: Spotted Knapweed (Centaurea maculosa). The Nature Conservancy
Muller, H., D. Schroeder, and A. Gassmann. 1988. Agapeta zoegana (L) (Lepidoptera: Cochylidae), a suitable prospect for biological control of spotted and diffuse knapweed, Centaurea maculosa Monnet De La Marck and Centaurea diffusa Monnet De La Marck (Compositae) in North America. Can. Ent. 120:109 24.
Rees, N.E., P.C. Quimby, Jr., G.L. Piper, E.M. Coombs, C.E. Turner, N.R. Spencer, and L.V. Knutson. 1996. Biological control of weeds in the west. Western Society of Weed Science, USDA Agric. Res. Serv., Mont. Dept. Agric., Mont. State Univ.
Roché, B.F. Jr., and C.J. Talbott. 1986. The collection history Centaurea found in Washington State. Agri. Res. Center. Res. Bull. XB0978. Washington State University Cooperative Roche, B.F., G.L. Piper, and C.J. Talbott. 1986. Knapweeds of Washington. Washington State Univ. Coop. Exten. EB1393. 41p. Extension, Pullman, WA. 36 pp.
Sheley, R.L., M. Manoukian, and G.Marks. 1996. Preventing Noxious Weed Invasion. Rangelands. 18(3):100 101.
Sheley, R.L., J.S. Jacobs, and M.F. Carpinelli. 1998. Distribution, biology, and management of diffuse (Centaurea diffusa) and spotted knapweed (Centaurea maculosa). Weed Technol. 12:353 362.
Sheley, R.L., J.S. Jacobs, and M.F. Carpinelli. 1999. Spotted knapweed. In: R.L. Sheley and J.K. Petroff (eds.), Biology and Management of Noxious Rangeland Weeds. Oregon State University Press, Corvallis, OR. pp. 350 61.
Shirman, R. 1981. Seed production and spring seedling establishment of diffuse and spotted knapweed. J. Range Manage. 34:45 47.
Spoon, C.W., H.R. Boweles, and A.Kulla 1983. Noxious weeds on The Lolo National Forest. A situation analysis staff paper. USDA Forest Ser. North. Reg. 33p.
Story, J.M., K.W. Boggs, W.R. Good, P. Harris, and R.M. Nowierski. 1991. Metzneria paucipunctella Zeller (Lepidoptera: Gelechiidae), a moth introduced against spotted knapweed: its feeding strategy and impact on two introduced Urophora spp. (Diptera: Tephritidae). Can. Entomol. 123:1001-1007.
Swearingen, J. M. 2003. Weed-US Database: Invasive Plants of Natural Areas. Plant Conservation Alliance. (draft)
Thompson, M.J. 1996. Winter foraging response of elk to spotted knapweed removal. Northwest Sci. 70(1):10 19.
Tyser, R.W. and C.H. Key. 1988. Spotted knapweed in natural area fescue grasslands: an ecological assessment. Northwest Sci. 62:151 160.
Virginia Department of Conservation and Recreation. 1999. Invasive Alien Plant Species of Virginia: Spotted knapweed (Centaurea maculosa Lam.). 2 p.

Whitson, T.D. et al. 2001. Weeds of the West. Western Society of Weed Science. 628 pp.

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Spotted knapweed
# *Centaurea maculosa* Larmarck or *C. biebersteinii* DC.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

The most cost effective management strategy for spotted knapweed is to prevent its spread to non-infested areas. Spread by seed can be minimized by avoiding travel through infested areas; by cleaning footwear, clothing, backpacks, and other items after hiking through infested areas; by not grazing livestock when ripe seeds are present in the flower heads; and by using weed free hay.

**Delaware River Invasive Plant Partnership - Fact Sheets - Spotted knapweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block, Morris Arboretum of the University of Pennsylvania - April 2002.**

Outlying plants should be controlled before main populations. The most effective way to control spotted knapweed is to begin a program when small patches of the plant appear. When patches are small, herbicide use is cost effective. Costs of herbicides for controlling knapweed may be prohibitory on large areas.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Centaurea maculosa - http://tncweeds.ucdavis.edu/esadocs/centmacu.html - Author(s): Teresa Mauer, Mary J Russo (Revision), Margaret Evans (Revision) - January 1987.**

Most literature on controlling knapweed has focused on reestablishing valuable range, pasture, or cropland. None has looked at the problem from the point of view of restoration ecology, with the intent of restoring the native community.

Spotted knapweed is increasing in its range and frequency in western North America. It is important to monitor whatever means of control are used in order to determine the efficacy of the efforts and the effects of control upon the larger community.

The extent of infestation can be monitored with either low altitude aerial photographs or permanent photo plots. A permanent line transect should be established so that plant and/or stem density can be measured. The transect should extend outside the colony in order to measure the direction and rate of change in the size of the colony. If biological control agents are used, sticky traps and capitula dissection should be used to monitor insect populations, attack rates, and seed losses.

There are several methods of control for this species. It is important to determine and document the methods most effective for different sized infestations, different communities, and the specific characteristics of the site, including soil type, exposure, drainage, and degree of disturbance, human or otherwise.

Control of this species is receiving considerable attention by state agencies as well as colleges and universities in Oregon, Washington, Idaho and Montana. This species is very aggressive. In addition to the effects it could have on elements, control of this species is mandated by county and state agencies. In most states and provinces it is under the "A" weed list for eradication.

Several grasses and forbs, most of them non-native, have been used to explore the possibility of replacing Centaurea species by the seeding of a competitor. A. H. Bawtree, Provincial Range Specialist in British Columbia cited a group of studies from which he recommended the application of picloram at no more than 6 oz. per acre followed by fall seeding of crested wheatgrass (Bawtree 1988). An Oregon State University study found six species--Palestine orchardgrass, Berber orchardgrass, Nangeela subterranean clover, Mt. Baker subterranean clover, and Covar sheep fescue--that over the course of six years were able to establish themselves and outcompete yellow starthistle (Centaurea solstitialis) (Johnson 1988). A two-year study of four grasses--Paiute orchardgrass, Covar sheep fescue, Critana thickspike wheatgrass, and Ephriam crested wheatgrass--found that the greater the biomass produced by the grass, the more it reduced the number of diffuse knapweed (Centaurea diffusa) seedlings. The species are listed in the order of their effectiveness. The researcher also indicated that those species whose growth period overlap the growth period of diffuse knapweed would be more effective at competing for moisture and nutrients (Larson, 1988).

Mowing is a method of control that would be possible only in areas that are not too rocky or steep, or without shrubs. If mowed in the early flowering state, the plants will usually regrow and produce abundant late season seeds. Those mowed even the same day as florets appear out of the bud have enough energy to produce seed. Among those mowed within ten days after flowerheads opened, none produced more than four filled seeds per head, and the greatest viability of these filled seeds was 57%, reached nine days after the flowerhead opened. Although these results indicate that mowing greatly reduces the seed set, a well established seed bank, such as would be present on a large or severe infestation, would most likely be able to compensate for this loss. Mowing would probably be a way to control populations, but not eradicate them.

No detailed research on vegetation response to knapweed control exists in the literature. The use of seeded or planted native bunchgrass species has not been explored. No studies have explored control by timed removal of flower capitula. Most studies have been designed to tackle infested areas on a large scale, and scale might prohibit removal of capitula by manual methods. However, relatively small areas that might be encountered on TNC preserves may be more amenable to this sort of management. Documented successful control in small areas by capitula removal would add valuable new information to the control literature.

Chemical and biological control have been proposed for spotted knapweed, and most of the control literature addresses these two categories:

Herbicides C. maculosa can be controlled with picloram (4 amino 3,5,6 trichloropicolinic acid) and 2,4 D but there are problems. Control by 2,4 D is temporary since it does not prevent germination from seeds in the soil. Picloram persists in soils but in 4 years, enough is lost from a .4 .6 kg/ha treatment to allow germination and reinfestation (Harris and Cranston 1979). The costs of applying picloram are estimated at $37/ha, and are prohibitive for very large infested areas (Maddox, 1979).

Other methods of control should be explored:

Mowing although this would not be feasible in rocky, or sagebrush areas, in some knapweed stands with little other vegetation it might be possible to mow the plants just after most flowering has ended but before seeds have matured. This would make regrowth unlikely since moisture levels late in the season are probably too low for continued growth, but would offer a possible advantage of reducing reserves for flowering the following year.

Hand Removal by August, in central and eastern Oregon, soils are often dry and dusty, and it may be possible to pull up a large number of seedlings, rosettes and reproducing plants in a small infested area. However, effects of soil disturbance on knapweed seed germination are not well documented. Even if seed germination of knapweed were not a problem, colonization by other weed species may be.

Hand Clipping This method might alleviate the soil disturbance problem outlined above. Again, this would probably be feasible only in small infested areas. Timing would be the same as mowing and the stems and capitula would be removed from the area. Again, control might be slower, due to continued emergence from seedbank reserves.

Burning although no literature specifically mentioned this as a control method for knapweed, it might be considered in areas with enough surrounding vegetation or litter to carry a controlled burn. Often however, dense stands of knapweed have little surrounding vegetation, possibly due to allelopathy. Litter from the previous year's stems often decays or scatters during the current season, but it may accumulate in very dense stands and create more favorable burning conditions.

**Wisconsin Department of Natural Resources - Non-native Plants Fact Sheets - Centaurea maculosa - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm**

Prevention is extremely important--spotted knapweed spreads readily in hay and on vehicle undercarriages. Caution is also necessary when using hay from the road ditches of primary roadways, or hay purchased from known infested areas. Outlying plants should be controlled before main populations.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

Small infestations of spotted knapweed can be controlled by persistent hand-pulling done prior to seed set. Gloves should be worn because of the possibility of skin irritation. Because spotted knapweed can regrow from the base, care must be taken to remove the entire crown and taproot.

**Delaware River Invasive Plant Partnership - Fact Sheets - Spotted knapweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block, Morris Arboretum of the University of Pennsylvania - April 2002.**

Small populations can be removed by digging or pulling. This is best done where the soil is moist. The entire root should be removed. Mowing has not been successful, plants merely reflower at a lower height. Established stands of knapweed may be controlled by hot prescribed burns followed by selective pulling and digging once the population has been reduced. Annual burns have reduced populations from 5-90 percent.

Reductions seem to correlate to the intensity of the burn administered; burns that remove nearly all the duff are most effective. Following a burn, it is important to reseed quickly with native species. The potential effects of intense burning on native species must be taken into consideration when planning a burn. Tillage that results in burying the seeds can be effective in preventing germination.

**Wisconsin Department of Natural Resources - Non-native Plants Fact Sheets - Centaurea maculosa - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm**

The most effective control is early detection and removal of pioneering plants. Small populations can be removed by digging or pulling. This is best done where the soil is moist. The entire root should be removed. Mowing has not been successful--plants merely reflower at a lower height.

## PRESCRIBED FIRE:

**The Nature Conservancy - Weed Notes: Spotted knapweed (Centaurea maculosa) - http://tncweeds.ucdavis.edu/esadocs/centmacu.html - TunyaLee Morisawa - June 1999.**

Hot prescribed burns may reduce established stands of knapweed. A follow-up of selective pulling and digging will further reduce populations. Annual burns have reduced populations by 5-90% and may be correlated with burn intensity. Reseeding with a native species is recommended. However, single, low intensity burns may actually worsen the problem since it is not hot enough to prevent resprouting and seed germination. Also, fires may disturb the area promoting colonization.

**Wisconsin Department of Natural Resources - Non-native Plants Fact Sheets - Centaurea maculosa - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm**

Once established, knapweed may be reduced by hot prescribed burns. These can be followed by selective pulling and digging once the population has been decreased. Annual burns have reduced populations anywhere from five to ninety percent. Reductions seem to correlate to the intensity of the burn administered; burns that remove nearly all the duff are most effective. Following a burn, reseed with native species. The potential effects of intense burning on native species must be taken into consideration when planning a burn.

## GRAZING:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

Long-term grazing by sheep and goats has been found to control spotted knapweed.

## BIOCONTROL:

**Van Driesche, R., et al., 2002, Biological Control of Invasive Plants in the Eastern United States, USDA Forest Service Publication FHTET-2002-04, 413 p. http://www.invasive.org/eastern/biocontrol/13Knapweed.html.**

Establishment and Spread of Agents
: Of the natural enemy species released against spotted knapweed in the eastern United States and Canada, the following have been established: Urophora affinis (Virginia, New York, Pennsylvania, Minnesota, Wisconsin, Michigan, Quebec); Urophora quadrifasciata (Connecticut, Indiana, Maryland, Massachusetts, Michigan, Minnesota, New Hampshire, New Jersey, New York, Pennsylvania, Rhode Island, Vermont, West Virginia, Wisconsin, and Quebec); Metzneria paucipunctella (Virginia); Agapeta zoegana (Minnesota); and Larinus minutus (Indiana, Minnesota) (Hoebeke, 1993; Wheeler, 1995; Wheeler and Stoops, 1996; Mays and Kok, 1996; Lang et al., 1997; Lang, pers. comm.) (Table 1). Except for the two Urophora species, particularly U. quadrifasciata, dispersal of the knapweed agents has been modest. An assessment of the Urophora spp. spread was conducted in Montana (Story et al., 1987).

Suppression of Target Weed:
Effects of imported natural enemies on spotted knapweed densities in the eastern United States have not been examined. However, impact information has been collected at sites in the western United States. Studies in Montana indicate that the two Urophora spp. are reducing spotted knapweed seed production by a minimum of 40% (Story et al., 1989), seed reduction by the Urophora spp. is further increased when M. paucipunctella is present (Story et al., 1991a), and A. zoegana is significantly reducing the biomass of spotted knapweed at some sites (Story et al., 2000). Spotted knapweed density is significantly reduced at two sites in western Montana where C. achates is well established (Story, unpub. data). Clark et al. (2001b) reported that spotted knapweed stem density, at 13 sites in Montana and adjacent states where U. affinis and one or both root feeding species of natural enemies (A. zoegana, C. achates) were established, declined from about 15 plants per m2 in 1991 to1993 to seven plants in 1997 to 1998. Effects on

Native Plants:
Neither the impact of these released agents on native, non-target plants, nor the recovery of native plant communities as weed densities, decline have been examined.

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

A variety of natural enemies are used as biological control agents for large infestations of spotted knapweed. Most biocontrol techniques use insect larvae to damage the root, stem, leaf, or flower. Two species of seed head flies, Urophora affinis and U. quadrifasciata, are well-established on spotted knapweed. The larvae of these species reduce seed production by as much as 50% by feeding on spotted knapweed seed heads and causing the plant to form galls. Three moth species (Agapeta zoegana, Pelochrista medullana, and Pterolonche inspersa) and a

weevil (Cyphocleonus achates) that feed on spotted knapweed roots have also been released. The collective stress on the plant caused by these insects reduces seed production and may lead to reduced competitiveness. Biological control agents may be more effective when combined with other control methods such as herbicides, grazing, and revegetation with desirable, competitive plants.

**Delaware River Invasive Plant Partnership - Fact Sheets - Spotted knapweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block, Morris Arboretum of the University of Pennsylvania - April 2002.**

In total, 14 insect and fungal species are presently being introduced or considered for introduction in North America to control spotted knapweed. Biological control agents include 2 root-mining moths, a flower moth, and a root-mining beetle. These have met with varying degrees of success. Most promising are 2 flies that attack the seed heads; Urophora affinis and U. quadrifasciata, have reduced seed production 95 percent in experimental populations. Both flies are being released experimentally in Wisconsin.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Centaurea maculosa - http://tncweeds.ucdavis.edu/esadocs/centmacu.html - Author(s): Teresa Mauer, Mary J Russo (Revision), Margaret Evans (Revision) - January 1987.**

Four insect species have been introduced into North America for biological control of knapweeds. Two gall flies, Urophora affinis and Urophora quadrifasciata (Diptera: Tephritidae) (Maddox 1982, Story and Anderson 1978, Harris 1980 a and b, Myers and Harris 1980, Berube 1980) and a moth, Metzneria paucipunctella (Lepidoptera: Gelechiidae) attack seed capitula (Englert 1971, Myers personal communication). A beetle which attacks the roots, Shenoptera jugoslavica (Coleoptera: Buprestidae) has also been introduced more recently (Zwolfer 1976).

U. affinis lays its eggs into young buds of C. maculosa. Egg hatch is synchronized with rapid growth of the receptacle in which each larva forms a gall. The seeds are not destroyed directly, but the diversion of nutrients to the gall reduces seed production by the plant as a whole (Harris 1980b). One generation of flies per season is usual, but a small proportion of the population completes a second one. Reported percentages of capitula attacked range from 10 50%, with up to 97% reduction in seed numbers per capitula.

U. quadrifasciata lays its eggs into florets inside more mature buds. Both species of Urophora can coexist in the same capitula. Some studies (Harris 1980) have found that U. quadrifasciata may attack capitula missed or more lightly attacked by the earlier attacking U. affinis, and result in a higher overall attack rate among capitula.

Metzneria paucipunctella lays eggs at the base of spotted knapweed buds and a young larva bores into the capitula after hatch. It feeds first on florets, then directly on seeds and does not form a gall. Establishment of this species has been somewhat difficult and increase has been slower than the fly species.

Female Sphenoptera beetles oviposit at the base of C. maculosa rosettes, and first instar larvae feed externally on plant tissues. After the first molt, the larva enters the plant tissue and mines into the root. A gall forms as the rosette terminates aestivation and resumes growth. The larva overwinters in the rosette root and pupates the next spring in a pupal chamber in the root crown. Adult beetles emerge and feed on knapweed leaves, adding to the root damage imposed by larvae.

**The Nature Conservancy - Weed Notes: Spotted knapweed (Centaurea maculosa) - http://tncweeds.ucdavis.edu/esadocs/centmacu.html - TunyaLee Morisawa - June 1999.**

Insects are available from USDA sources and could be released in target areas as a first step for control with relative ease and at no or little cost. Seed capitula attack percentages seem to rise quickly within a few years, but noticeable decreases in reproductively mature plants will take longer because of seed bank reserves and dormancy. This method, though slower, may be desirable because of minimal disturbance to soil and surrounding vegetation.

In addition to the biocontrol agents listed in the ESA, a seedhead weevil, Bangasternus fausti (Reitter), that is native to Europe, was released in the US in 1991 for the control of spotted knapweed, diffuse knapweed (C. diffusa) and purple star-thistle (C. calcitrapa). The weevil has become established in several states: Montana, Nebraska and Oregon. It has not been shown to reduce populations or even significantly slow their spread. While the larvae can destroy 100 percent of the seeds in a seedhead, not all seedheads are affected and so seed production is still high. Many seeds that do develop may not germinate. TNC has not used this biocontrol and gaining approval would be a formidable task.

**Wisconsin Department of Natural Resources - Non-native Plants Fact Sheets - Centaurea maculosa - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm**

Several biological controls exist, including two root mining moths, a flower moth, and a root mining beetle. These have met with varying degrees of success. Most promising are the two seed-head attacking flies Urophora affinis and U. quadrifasciata. Congruently, these two flies have reduced seed production 95% in experimental populations. While both flies are being released experimentally in Wisconsin, their effectiveness here is still unknown. These insects were not available for general use at the time of printing. The USDA should be consulted to determine the current status of these controls.

## CHEMICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

Control of spotted knapweed infestations using three chemical herbicides (2,4-D, clopyralid, and picloram) has been reported but is problematic. Existing plants can be killed with 2,4-D but it needs to be reapplied yearly to control new plants germinating from seed stored in the soil. Picloram is a more persistent herbicide and has controlled knapweed for three to five years when applied at 0.25 lb/acre at any stage of plant growth; or with clopyralid (0.24 lb/acre) or clopyralid (0.2 lb/acre) plus 2,4-D (1 lb./acre) applied during bolt or bud growth stage. In the absence of desirable

native grasses, longevity of control may be increased by revegetating with competitive grasses and forbs. Picloram may pose a risk of groundwater contamination where soils are permeable, particularly where the water table is shallow.

**Delaware River Invasive Plant Partnership - Fact Sheets - Spotted knapweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block, Morris Arboretum of the University of Pennsylvania - April 2002.**

Chemical controls are an effective means of eliminating spotted knapweed. A 3% solution of triclopyr in the water-soluble formulation has been found to be effective. This application should be repeated 3-4 times per year for two years to control plants growing from the seed bank. Triclopyr will not affect grasses. Experimentation to test the effectiveness of glyphosate is needed.

**The Nature Conservancy - Weed Notes: Spotted knapweed (Centaurea maculosa) - http://tncweeds.ucdavis.edu/esadocs/centmacu.html**

Clopyralid (Transline) applied at 0.13-0.19 L/ha (0.66-1.0 pt/acre) or clopyralid + 1.12 kg/ha 2,4-D (Curtail) provide control with little soil residual. Apply during the bolt or bud stage. Less control was achieved when applied to the rosette, flowering and after flowering stages.

Picloram (Tordon) (0.28 kg/ha), as listed in the ESA for C. maculosa, can control plants and seedling for 2-3 years. However, as mentioned in the ESA, there is a long residual and it is costly (as is dicamba - listed below). The residual may be shorter on gravel soils, wet areas and areas with high soil organic matter. The optimum time for the application of picloram is during the rosette growth stage in the fall or in the bud to bloom stage in the spring. Picloram can not be used near water or in porous substrata overlying ground water. Picloram does not affect grasses, but long term affects have been observed from it on shrubs and trees, possibly due to it leaching in ground water.

Dicamba (Banvel) will also provide control of plants and shorter residual control of seedlings at a rate of 0.18-0.37 kg/ha (1-2 lbs/acre). A follow-up treatment of 2,4-D at 0.18 kg/ha (1 lb/acre) annually may be needed to prevent reinfestation.

2,4-D is listed in the ESA for control, however, after stem elongation it should not be applied since control is not very effective. There is no residual control and so annual applications are necessary.

Triclopyr (water-soluble formulation) applied at the rate of 3% a.i. can be sprayed on the plant (except the flower) 3-4 times a year for control. Triclopyr does not affect grasses.

For all chemical applications treat the area around C. maculosa patches (3-4.5 m). Follow-up treatments are extremely important for the continual control of spotted knapweed.

**Wisconsin Department of Natural Resources - Non-native Plants Fact Sheets - Centaurea maculosa - http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm**

Chemical controls are an effective means of eliminating spotted knapweed, but they may also have the most adverse consequences. The recommendations from western rangelands involve the use of some very potent chemicals. These restricted chemicals are not recommended for use on quality natural areas, but may be appropriate on roadsides and other highly disturbed areas. Experimentation testing the effectiveness of less toxic broad-leaf herbicides such as triclopyr or glyphosate is encouraged.

A 3% active ingredient (a.i.) solution of triclopyr in the water-soluble formulation with dye can be sprayed on the entire plant except the flower, which should be spared for native fauna. This application should be repeated 3-4 times per year for two years. Triclopyr will not affect grasses. Picloram at 0.25 to 0.5 pounds per acre will control spotted knapweed plants and seedlings for 2-3 years, although the residual control period may be shorter on gravel soil, where soil organic matter is high, or in wet areas. Picloram should be applied either in fall when the plant is in the rosette growth stage, or in spring during the bud to bloom stage. Picloram should not be used near water or on sandy soils with ground water ten feet or less below the surface. Dicamba at 1-2 pounds (acid equivalent--see label) per acre also controls spotted knapweed, but may require annual follow-up treatment for a minimum of two years.

Clopyralid is more selective, affecting only legumes and composites. It can be applied at 0.25 pound acid equivalent per acre. A mixture of 0.19 pounds of clopyralid per acre plus 2,4-D at 1 pound acid equivalent per acre is also an option; both herbicides provide good control of spotted knapweed with less soil residual than picloram or dicamba. Spotted knapweed that is still in the rosette stage can be controlled by applying a 2,4-D low volatile ester, oil soluble amine, or water soluble amine formulations at 2 pounds per acre. Annual spraying for several years may be required to deplete the seed bank.

Picloram and clopyralid are the most commonly used and effective herbicides for spotted knapweed. Picloram causes the largest initial decrease in native forb cover, but the experiment that produced this data also concluded that most non-target forb populations were tolerant of herbicidal treatments and benefited from being released from knapweed competition after three years of selective herbicidal application. Application of these herbicides has not been found to decrease the frequency of occurrence of grasses, sedges, shrubs, or trees in the treated site. Herbicides were applied manually using a carbon dioxide pressure-regulated backpack sprayer at the concentrations listed in previous paragraphs.

## OTHER CONSIDERATIONS:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Spotted knapweed- http://www.nps.gov/plants/alien/fact/cebi1.htm - Author(s): Michael Carpinelli - May 1, 2003.**

Burning, cultivation, and fertilization typically are not effective on spotted knapweed unless combined with other methods of control.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Greater celandine
## *Chelidonium majus* L.

**SOURCE**: The following information on this species is taken from the Invasive Plant Atlas of New England (IPANE) website at http://invasives.eeb.uconn.edu/ipane/. Modifications include additional resource information. Check the website link for future updates.

**DESCRIPTION:** *Chelidonium majus* is a brittle, herbaceous biennial that reaches 30-80 cm (11-31 in.) in height. Its stems are ribbed and branching. The lower parts of the branches are pubescent. If the branches or leaves are broken, yellowish-orange sap can be seen. The alternate cauline leaves can be up to 35 cm (13 in.) in length, with petioles measuring 2-10 cm (0.75-4 in.) long. The thin leaf blades are glaucous beneath, deeply 5-9 lobed and are irregularly dentate around the margins. The veins of the lower leaf surfaces have fine, short hairs.

The bright yellow flowers of *Chelidonium majus* are contained in axillary pedunculate umbels. The peduncle itself measures 2-10 cm (0.75-4in.) long. Each flower has four obovate to oblong petals that measure about 1 cm (0.4in.) wide. This plant usually flowers from May to June. The "lumpy" capsules are linear to oblong-shaped and plant measure 2-5 cm (0.75-2 in.) in length. Within the capsule are black seeds with reticulate pitting on their surface.

Page References Bailey 428, Fernald 680, Flora of North America 302, Gleason and Cronquist 67, Holmgren 64, Magee & Ahles 526, Newcomb 142, Peterson & McKenny 130. See reference section below for full citations.







Photos: Top to Bottom (#1-5):
#1 and 4: Stacey Leicht, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/
#2, 3, 5: Leslie J. Mehrhoff, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/

**SIMILAR SPECIES:** *Glaucium flavum* Crantz. (Yellow hornpoppy), *Stylophorum diphyllum* (Michx.) Nutt (Celandine poppy), *Cardamine impatiens* L. (Bushy rock-cress),*S. diphyllum* is not-native to New England (it is native futher south), but is often cultivated. Basal rosettes of *C. impatiens* could be confused for *C. majus*. The rosettes of *C. majus* are pubescent, while those of *C. impatiens* are glabrous.

**REPRODUCTIVE/DISPERSAL MECHANISMS:** The seeds of this plant are ant-dispersed. This movement is facilitated by an elaiosome on the seed which attracts the ants so that they move it.

**DISTRIBUTION:** *Chelidonium majus* is native to Eurasia from Spain to Turkey and Russia, north to Great Britain and Ireland and south to North Africa. In the United States it is located from Maine to Georgia and west to Minnesota, Iowa and Missouri. There are also reports of it in Washington, Montana and Utah. This plant has been reported from all the states of New England.

**HISTORY OF INTRODUCTION IN NEW ENGLAND:** *Chelidonium majus* was most likely introduced into New England as an herbal remedy for skin diseases. It was reported to be in New England herb gardens as early as 1672. From these gardens the plant became naturalized and spread across the landscape.

**HABITATS (IN NEW ENGLAND):** Early Successional Forest, Edge, Floodplain Forest, Open Disturbed Area, Roadside, Vacant Lot, Wet Meadow, Yard or Garden. *Chelidonium majus* is most often found in disturbed areas especially with moist soil. This plant could be moved conceivably wherever there are ants to move the seeds.

**THREATS:** *Chelidonium majus* can become abundant in minimally managed situations, and can outcompete other native herbaceous plants.

**For more information on greater celandine, contact:**
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA - NRCS PLANTS Database, http://plants.usda.gov/

**REFERENCES:**
Bailey, L. H. 1949. Manual of Cultivated Plants. Macmillan, New York.
Fernald, M.L. 1950. Gray's Manual of Botany 8th edition. American Book Company, New York. Flora of North America Editorial Committee, eds. 1993+. Flora of North America North of Mexico. Volume #3. Oxford University Press
Gleason, H. A. 1952. The New Britton and Brown Illustrated Flora of the Northeastern United States and Adjacent Canada. Macmillan Publishing

Co., Inc. New York
Gleason H.A. and A.C. Cronquist. 1991. Manual of Vascular Plants of the Northeastern United States and Adjacent Canada. 2nd ed. New York Botanical Garden, Bronx, New York.
Holm, L.G., Pancho, J.V., Herberger, J.P., and Plucknett, D.L. (1979). A Geographical Atlas of World Weeds. John Wiley and Sons, New York, USA.
Holmgren N.H. 1998. Illustrated Companion to Gleason and Cronquist's Manual. New York Botanical Garden, Bronx, New York.
Josselyn, John. 1672. New England Rarities Discovered. Meriden Gravure Company, Meriden, Connecitcut.
Magee D.W and H.E. Ahles. 1999. Flora of the Northeast. University of Massachusetts Press, Amherst.
Newcomb N. 1977. Newcomb's Wildflower Guide. Little Brown, Boston.
Peterson R.T. and M. McKenny. 1968. A field Guide to Wildflowers of Northeastern and North-central North America. Houghton Mifflin, Boston.
USDA, NRCS. 2001. The PLANTS Database, Version 3.1. (http://plants.usda.gov). National Plant Data Center, Baton Rouge, LA 70874-4490 USA.

**Management and Control Information**

Return to List




## Species Management and Control Information

### Greater celandine
### *Chelidonium majus* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

**Management information for this species was unavailable at time of tutorial development**

Return to List




# Canada thistle
## *Cirsium arvense* (L.) Scop.

**SOURCE**: The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org and http://www.ppws.vt.edu/weedindex.htm) and resource information. Check the website links for future updates. The last update for this fact sheet is March 28, 2005.

**NATIVE RANGE:** Temperate regions of Eurasia
**DESCRIPTION:** Canada thistle is an herbaceous perennial with erect stems 1½-4 feet tall, prickly leaves and an extensive creeping rootstock. Stems are branched, often slightly hairy, and ridged. Leaves are lance-shaped, irregularly lobed with spiny, toothed margins and are borne singly and alternately along the stem. Rose-purple, lavender, or sometimes white flower heads appear from June through October, generally, and occur in rounded, umbrella-shaped clusters.



Photo: John M. Randall,
The Nature Conservancy, Davis, CA




Photo: Chris Evans, The University of Georgia,
www.forestryimages.org

Photo: Virginia Tech Weed Guide,
http://www.ppws.vt.edu/weedindex.htm



The small, dry, single-seeded fruits of Canada thistle, called achenes, are 1-1½ inches long and have a feathery structure attached to the seed base. Many native species of thistle occur in the U.S., some of which are rare. Because of the possibility of confusion with native species, Canada thistle should be accurately identified before any control is attempted.

**ECOLOGICAL THREAT:** Natural communities that are threatened by Canada thistle include non-forested plant communities such as prairies, barrens, savannas, glades, sand dunes, fields and meadows that have been impacted by disturbance. As it establishes itself in an area, Canada thistle crowds out and replaces native plants, changes the structure and species composition of natural plant communities and reduces plant and animal diversity. This highly invasive thistle prevents the coexistence of other plant species through shading, competition for soil resources and possibly through the release of chemical toxins poisonous to other plants.



Photo: ©John M. Randall,
The Nature Conservancy,
Davis, CA.

Canada thistle is declared a "noxious weed" throughout the U.S. and has long been recognized as a major agricultural pest, costing tens of millions of dollars in direct crop losses annually and additional millions costs for control. Only recently have the harmful impacts of Canada thistle to native species and natural ecosystems received notable attention.

**DISTRIBUTION IN THE UNITED STATES:** Canada thistle is distributed throughout the northern U.S., from northern California to Maine and southward to Virginia. It is also found in Canada, for which it was named. Canada thistle has been identified as a management problem on many national parks and on preserves of The Nature Conservancy in the upper Midwest, Plains states, and the Pacific northwest.

**HABITAT IN THE UNITED STATES:** Canada thistle grows in barrens, glades, meadows, prairies, fields, pastures, and waste places. It does best in disturbed upland areas but also invades wet areas with fluctuating water levels such as streambank sedge meadows and wet prairies.

**BACKGROUND:** Canada thistle was introduced to the United States, probably by accident, in the early 1600s and, by 1954, had been declared a noxious weed in forty three states. In Canada and the U.S., it is considered one of the most tenacious and economically important agricultural weeds, but only in recent years has it been recognized as a problem in natural areas.

**BIOLOGY & SPREAD:** Canada thistle produces an abundance of bristly-plumed seeds which are easily dispersed by the wind. Most of the seeds germinate within a year, but some may remain viable in the soil for up to twenty years or more. Vegetative reproduction in Canada thistle is aided by a fibrous taproot capable of sending out lateral roots as deep as 3 feet below ground, and from which shoots sprout up at frequent intervals. It also readily regenerates from root fragments less than an inch in length.

**AUTHORS:**
Gwendolyn Thunhorst, The Nature Conservancy, Arlington, VA.
Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on Canada thistle, please contact:**
Cornell University, Biological Control of Weeds-Canada thistle, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Daniel Roddy, U.S. National Park Service, Wind Cave National Park, SD, dan_roddy@nps.gov Illinois
Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Invasive Plants of Ohio Fact Sheets, http://www.dnr.state.oh.us/dnap/invasive/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild, http://www.nps.gov/plants/alien/factmain.htm.
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA Forest Service, Northeastern Area, http://www.na.fs.fed.us/fhp/invasive_plants/weeds.
USDA-NRCS PLANTS Database, http://plants.usda.gov/
U.S. Geological Survey, Biological Resources Division, Flagstaff, AZ
http://www.nbs.nau.edu/FNF/Vegetation/Exotics/Cirsium/cirsiumarvense.html
Virginia Natural Heritage Program Fact Sheet-Canada thistle; http://www.dcr.virginia.gov/dnh/invlist.htm
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm

**REFERENCES:**
Evans, J.E. 1984. Canada thistle (Cirsium arvense): a literature review of management practices. Natural Areas Journal 4(2):11-21.
Hutchison, M. 1992. Vegetation management guideline: Canada thistle (Cirsium arvense (L.) Scop.). Natural Areas Journal 12(3):160-161.
The Nature Conservancy. Canada Thistle: Element Stewardship Abstract. In: Wildland Weeds Management & Research Program, Weeds on the Web.

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Canada thistle
# *Cirsium arvense* (L.) Scop.

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance - Alien Plant Working Group - WeedS Gone Wild - Fact sheet - Canada thistle - http://www.nps.gov/plants/alien/fact/ciar1.htm - Authors: Gwendolyn Thunhorst and Jil M. Swearingen - March 2005.**

Management of Canada thistle can be achieved through hand-cutting, mowing, controlled burning, and chemical means, depending on the level of infestation and the type of area being managed. Due to its perennial nature, entire plants must be killed in order to prevent regrowth from rootstock. Hand-cutting of individual plants or mowing of larger infestations should be conducted prior to seed set and must be repeated until the starch reserves in the roots are exhausted. Because early season burning of Canada thistle can stimulate its growth and flowering, controlled burns should be carried out late in the growing season for best effect.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

Management Requirements:

*Cirsium arvense* should be removed from high quality natural areas when it is first observed. The plant is very tenacious and difficult to control once established. In lower quality areas, management effort should be influenced by the extent of invasion; greater effort is warranted in areas that have new and/or small invasions which are more likely to be eliminated or contained.

Management Programs:
*Cirsium arvense* management programs should be designed to kill established clones since the species spreads primarily by vegetative expansion of the root system. Prevention of seed production is a secondary consideration since spread by seeds is relatively rare. On the other hand, seedlings are the most susceptible growth stage (Bakker 1960). In areas that are susceptible to thistle invasion but which have not yet been invaded, management programs should be implemented to prevent the species from becoming established.

It is important to understand the biology of *Cirsium arvense* as control is greatly influenced by clonal structure (Donald 1994), growth stage (Tworkoski 1992), season of treatment, weather conditions, ecotype (Hodgson 1964), soil type, and control method(s) used. A single control method is rarely effective and it is often necessary to use two or more methods at any given site (Lee 1952, Donald 1992, Diamond 1993). In addition, treatments or combinations that are effective at one site may be ineffective at others (Frank and Tworkoski 1994).

It takes at least two growing seasons to determine whether a particular control method is effective. Several studies have recorded a temporary decline in *Cirsium arvense* in the first year after treatment, followed by a return to the pre-treatment conditions the second growing season (Zimdahl and Foster 1993).

The literature on Canada thistle control focuses on agricultural systems. Management in natural areas is more difficult due to the need to protect native species and communities. At this time, there are no control methods suitable for wide-spread use in natural areas that eradicate, rather than reduce, Canada thistle.

Management strategies should be adjusted to reflect weather conditions. For example, drought stress reduces the effectiveness of most herbicides against *Cirsium arvense* (Haderlie et al. 1987), but increases the effectiveness of mechanical controls (Hansen 1918, Johnson 1912). Thus, mowing or burning would be preferred strategies under drought conditions.

## MANUAL AND MECHANICAL CONTROL:

**Illinois Nature Preserves Commission - Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/cthistle.html - Vol.1, No. 7 - Author: Max Hutchison - February 6, 1990.**

Repeated and frequent pulling or hand-cutting of individual plants will eventually starve underground stems. Cutting or pulling should be at least 3 times each season, in June, August, and September. This treatment is feasible for light and moderate infestations, but may be relatively time consuming in heavy infestations.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

MANIPULATION OF WATER LEVEL AND SALINITY:
There is no information on the impacts of manipulating water levels on *Cirsium arvense*, and little on impacts of manipulating soil salinity. Salt was one of the earliest chemicals used to kill *Cirsium arvense*. Applications of 180-640 pounds/acre used in the 1920's killed *Cirsium arvense* (Hodgson 1968). but the species is tolerant of lower salt concentrations. Seed germination is reduced but not prevented by modest concentrations of NaCl; Wilson (1979) recorded germination rates of 83% without NaCL, 67% at 10,000 ppm NaCl, and 14% at 20,000 ppm NaCl (seawater is about 35,000 ppm salt).

MOWING, DISCING AND PULLING:
Mowing temporarily reduces above-ground biomass, but does not kill *Cirsium arvense* unless repeated at 7-28 day intervals for up to 4 years. This intensity of mowing is not recommended in natural areas, where it would likely damage native vegetation. Mowing just twice a year, in mid-June and September may reduce or contain Canada thistle. When mowing, cut high enough to leave > 9 leaves/stem, or >20 cm of bare stem tissue, as mature Canada thistle leaves and stems independently inhibit development of shoots from rootbuds. When the primary stem is removed, rootbuds are stimulated to produce new shoots that might otherwise be suppressed, especially under low humidity. Under high humidity, root buds are stimulated to develop shoots regardless of presence of stem or leaves (Hunter et al. 1985). Cut plants also produce twice the length and weight of new shoots after just seven days under high humidity (100%) than low humidity (50%; Hunter et al. 1985).

Early studies recommended mowing at frequent intervals to starve Canada thistle's root systems and remove it from farm fields and pastures (Cox 1913, Johnson 1912, Hansen 1918, Detmers 1929). Mowing monthly for a four-year period eliminated practically all thistles (Welton et al. 1929) and mowing at 21-day intervals weakened roots and prevented seed production (Seely 1952). Hodgson (1968) found that mowing alfalfa fields twice annually, at Canada thistle's early-bud to pre?flowering stage (early to mid-June in Montana) and early fall (September) reduced Canada thistle to 1% of its initial value in four years. Mowing two to three times a year can prevent seed set (Hansen 1913, Rogers 1928) but mowing once a year is ineffective (Donald 1990). In order to prevent production of viable seeds, stems must be mown before the flowers open when they have been open for only a few days. Stems with flowers that have been open 8-10 days can develop viable seeds (Derscheid and Schultz 1960).

Tilling can reduce or eliminate *Cirsium arvense*, if conducted repeatedly for several years. Tilling 7-10 cm deep every 21 days (6 cultivations over the growing season) reduced *Cirsium arvense* shoots by 98% in Montana (Hodgson 1958), and "eradicated" *Cirsium arvense* in Idaho (McKay et al. 1959). Ecotypes of *Cirsium arvense* differ somewhat in their response to repeated cultivation, but all were controlled by 10 cultivations over 1 1/2 growing seasons (Hodgson 1970). Tilling is not recommended in natural areas, however, because it would severely damage native vegetation and tilling can sometimes spread Canada thistle across and between fields (Willard and Lewis 1939).

Tilling affects only the upper part of the root system and in some cases as little as a quarter of Canada thistles roots are in the top 20 cm of soil reached by normal tillage while the majority of roots are 20-40 cm deep and some roots reach to 1.8 m deep (Nadeau and Vanden Born 1989). New shoots develop after tilling. Root fragments from a single young plant can produce over 900 shoots when the roots are cut into 10 cm fragments (Nadeau and Vanden Born 1989), as typically occurs with discing. Deep tilling (10-20 cm below the soil surface) is more effective than shallow tilling (surface), Fewer shoots are present 40 days after deep tilling and shoots cut at depth require more time to emerge than shoots cut near the surface (Darwent et al. 1994).

When tilling is discontinued after early August, new shoots do not produce flower stalks. Repeated tillage, however, kills *Cirsium arvense* by preventing shoot growth and thus depleting roots and their fragments of nutrient reserves (Donald 1990). Leaving large clods (5.3 cm diameter) minimizes seed germination and leaving small clods (1.5 cm diameter) can stimulate germination of seedling which can be killed by retilling or treating with herbicide (Terpstra 1986). Seedlings should be disced or treated with herbicide within 2 1/2 weeks of thistle germination because after that time they will have developed roots that can survive discing and some herbicide treatments (Haderlie et al. 1987). Eight percent of seedlings (19 days old) with 2 true leaves resprouted when their tops were cut (Wilson 1979).

In Canada the most successful control method for *Cirsium arvense* is the "August rosette method", consisting of tilling until mid to late-July, applying herbicide in mid-August, and tilling again after 3 weeks (Alberta Agriculture 1993, Saskatchewan Agriculture and Food 1993, cited in Darwent et al. 1994b, Hunter 1996). Darwent et al. (1994b) recommend tilling *Cirsium arvense* patches until August 1, waiting 40 days for all shoots to emerge, and then applying glyphosate to the new shoots. Tilling until August 1 ensures that newly emerged shoots will remain as rosettes, as *Cirsium arvense* flowers only under long-days. Waiting 40 days is necessary to obtain adequate shoot emergence, and for shoots to grow large enough for effective glyphosate activity (Darwent et al. 1994b). This method is not practical in most natural areas unless thistles are mown or individually cut near the soil surface instead of tilled.

Reversing this procedure (applying herbicide and then tilling or discing) is ineffective regardless of herbicide type, season of herbicide application, or time between discing and herbicide treatment (Zimdahl and Foster 1993). Destroying shoots by discing releases dormant buds, and may increase the total number of shoots (Zimdahl and Foster 1993). A minimum of 3 days between glyphosate and tilling is needed for glyphosate to damage root system (Carlson and Donald 1988); waiting longer may further increase thistle mortality.

Discing in Mid-June is ineffective, as cut stems readily develop new roots and establish new clones (Magnusson et al. 1987). Fewer cut stems survive when discing is conducted in mid-September, and surviving stems do not develop adequate root systems to survive the winter (Magnusson et al. 1987). These roots are also more likely to be winterkilled, since disced fields accumulate less snow cover than undisced fields, and soil temperatures are lower (*Cirsium arvense* roots are injured at -2 degrees C and killed at -8 degrees C; Schimming and Messersmith 1988). Discing an all-male population may result in development of female plants.

SMOTHERING:
Mulching is impractical. Manure must be spread 2 m thick and cover an area 5-6 m in diameter *Cirsium arvense*, and plants that emerge at the mulch perimeter must be removed. Likewise, mulching with hay is ineffective, as roots extend beyond the covered shoots (Willard and Lewis 1939). Mulching may actually enhance *Cirsium arvense* overwinter survival, as mulch insulates cold-sensitive roots.

However, covering Canada thistle with boards, sheet metal or tar paper can kill the plants (Spence and Hulbert 1935).

COMPETITION:
Smother crops may be grown to choke and shade out undesirable species. To be effective against Canada thistle the crop must come up first, grow rapidly during the early summer in order to shade out the thistle, and retain vigor until frost (Rogers 1928). These principles apply in the selection of smother crops on cultivated fields or haylands, but may also be applicable in the selection of native "smother" species in prairie restorations.

Smother crops are used in integrated pest management systems for Canada thistle on agricultural lands (Hodgson 1968) but the smother crops known to be effective are themselves invasive. Alfalfa (*Medicago sativa*) and especially sweet clover (*Melilotus alba* and/or *M. officinalis*) compete with and can reduce spread of *Cirsium arvense*, particularly when mowed as haycrops 3 times/year (Detmers 1927, Rogers 1928). However, these species are persistent and/or invasive in natural areas. Competition from timothy, orchard grass or redtop is ineffective (Detmers 1927).

Competition from tall fescue (*Festuca arundinacea*), in combination with the defoliating beetle *Cassida rubignosa*, reduced *Cirsium arvense* density after three years, but not after two years (Ang 1993). Competition from tall fescue was more detrimental to *Cirsium arvense* than competition from crownvetch (*Coronilla varia*) (Ang et al. 1995), and damage increased in when tall fescue was used in combination with *Cassida rubignosa*.

ENVIRONMENTAL:
Control efforts may be more successful when *Cirsium arvense* is under environmental stress. The plant is drought and flood sensitive, and its roots are cold-sensitive. Cutting or applying herbicide to shoots after a very severe winter may add sufficient stress to kill plants.

## PRESCRIBED FIRE:

**Illinois Nature Preserves Commission - Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/cthistle.html - Vol.1, No. 7 - Author: Max Hutchison - February 6, 1990.**

Prescribed fire can be effective in controlling this species and is a preferred treatment. Late spring burns, between May and June, are most detrimental to this noxious weed and should be used when possible. Prescribed burns to control this plant should not be conducted early in the spring, as early spring burns can increase sprouting and reproduction of this species. During the first 3 years of control efforts, burns should be conducted annually.

Management practices that maintain and encourage the development of healthy stands of native species will help prevent establishment of Canada thistle or help shade and weaken plants on sites already infested.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

*Cirsium arvense* response to fire varies from positive to negative, depending on season of burn, soil moisture, and location. Dormant season burning stimulates growth of native herbaceous species which compete with Canada thistle. Growing season fire damages native species as well as Canada thistle.

In a mesic grassland in Oregon, dormant season fire reduced Canada thistle flowerhead and seed production. Flowering plants had equal density in burned and unburned plots (55-61/m2) but produced 50% fewer flowerheads in the burned plots (18/shoot vs 36/shoot, respectively; Young 1986). Additionally, plants in the burned plots produced an average of 1.2 "functional" or seed-producing flowerheads/shoot, compared to 16.3/shoot in the unburned plots. Dormant season burning (December or April) also stimulated production of numerous small Canada thistle shoots, resulting in higher density but equal biomass (Young 1986).

In North Dakota, dormant season burning reduced the relative abundance of Cirsium arvense by stimulating growth of native vegetation (Carlson 1987). Growing season fires reduced thistle density but harmed native species (Smith 1985). Plots burned in mid-June had heavy seed production July through September, while plots burned mid-July to mid-August had numerous seedlings in August and September but they failed to survive the winter (Smith 1985).

In Alberta, Canada, spring burning in a marsh favored growth of native species and did not alter *Cirsium arvense* biomass (Thompson and Shay 1989). An August fire increased biomass and shoot density of *Cirsium arvense*, which averaged 63 g/m2 vs 5 g/m2, and 20 shoots/m2 vs 0.9 shoots/m2, on burned and unburned plots, respectively (Thompson and Shay 1989). Seedling density also increased following the summer fire. In another wetland in Alberta, Canada, *Cirsium arvense* cover was not affected by fire (Hogenbirk and Wein 1991), but increased when the wetland area was subjected to drought.

## GRAZING:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

Very young plants are eaten by goats or sheep in the spring, but grazing is the least effective control method for Canada thistle (Rogers 1928). Cattle and horses avoid *Cirsium arvense*, and browse on competing vegetation, which results in gradual dominance by *Cirsium arvense*. Heavy grazing breaks up sod, which permits seeding in of Canada thistle.

There are no available data on the effect of stocking rates or grazing intensities on Canada thistle. It seems likely that animal disturbance from conventional grazing encourages the spread of Canada thistle, as has been demonstrated for *C. lanceolatum*, *C. vulgare*, and *C. undulatum* (Tomarek and Albertson 1953, Ankle 1963, Hetzer and McGregor 1951).

## BIOCONTROL:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

Overall, biocontrol currently provides little or no control of Canada thistle populations, although some agents weaken and kill individual plants. Most potential biocontrol organisms are not adequately synchronized with *Cirsium arvense*'s life cycle in North America to induce high mortality. Management that delays *Cirsium arvense* maturation, such as mowing or burning, may help synchronize the susceptible thistle growth stage to the biocontrol agent life cycle (Forsyth and Watson 1985a).

*Cirsium arvense* has few or no effective natural enemies in its native habitat, where it is also considered a severe agricultural weed (Peschken 1971). In all, more than 130 species, including diseases, birds, and >80 insects, attack Canada thistle (Maw 1976). At any one site in its native range, however, an average of 4.5 insect species attack *Cirsium arvense* but in general they cause little damage, as their densities are usually low and most species consume little plant material (Freese 1995). In North America, larvae of the native painted lady butterfly (*Vanessa cardui*; Lepidoptera) feed on Canada thistle and other related thistles and cause extensive defoliation within localized areas, but impact varies year to year due to migration patterns (Story et al. 1985).

At least 7 insect species have been intentionally or unintentionally released for Canada thistle control in North America and a few of them cause conspicuous damage. The beetle *Cassida rubiginosa* was introduced accidentally in 1902 and defoliates plants (Maw 1976). Larvae of the intentionally introduced biocontrol weevil *Ceutorhynchus litura* feed on stems of Canada thistle. The introduced stem-galling fly *Urophora cardui* attacks thistle shoots but has little impact. Larvae of the fly *Orellia ruficauda* (Diptera) damage seed heads. The beetles *Altica carduorum* and *Lema cyanella* feed on Canada thistle's leaves. The seed weevil *Rhinocyllus conicus* was introduced to control musk thistle (*Carduus nutans*) and other related *Carduus* and *Cirsium* thistles and lays eggs in Canada thistle flowerheads. The weevil *Larinus planus* is a seed head feeder but it has had little impact on Canada thistle and attacks native thistles. Two pathogens have also been considered for use against Canada thistle. The rust *Puccinia punctiformis* and the fungus *Sclerotinia sclerotiorum* attack shoots and roots respectively. Of all these biocontrol organisms, *Orellia ruficauda* and *Puccinia punctiformis* appear to inflict the most significant damage (Maw 1976, Forsyth and Watson 1985a), but even this is probably not sufficient to control Canada thistle populations.

A combination of biocontrol agents, or of biocontrol agents and herbicides, may provide better control of Canada thistle than any single agent. It has been suggested that at least three biocontrol organisms may be needed for effective Canada thistle control (Forsyth and Watson 1985a) In western Canada, where Cassida rubiginosa and Puccinia punctiformis do not occur, Cirsium arvense is a much greater problem than in the eastern part of Canada, where these organisms are present. In Ontario there appeared to be a synergistic relationship between infestation of thistle by Ceutorhynchus litura and infection by the rust Puccinia punctiformis. 87% of rust-infected thistles were mined by weevils compared with 32% of uninfected shoots (Peschken and Beecher 1973). Such an effect is not reported for sites in western Canada (Peschken and Wilkinson 1981). Impacts of Ceutorhynchus litura are also enhanced when Canada thistle is infected with the fungal pathogen Sclerotinia sclerotiorum. Ceutorhynchus mining may have significant impacts after Sclerotinia infection under drought conditions, especially in the western Great Plains (Bourdot et al. 1995). Vegetative shoots were most susceptible to the disease (Bourdot et al. 1995) but the disease was not transferred to shoots that emerged after the initial infection. This disease is therefore most effective as a control method if applied after the majority of shoots have emerged. Disease development, however, requires high moisture conditions which are less likely as the growing season progresses in most areas (Bourdot et al. 1995). Thus, timing of application is critical, and varies between sites and years.

Schroeder (1980) suggested that a combination of root- and shoot-feeding insects will be needed for effective biological control but no root feeders are known that cause substantial damage to Cirsium arvense (Ang et al. 1995). The organisms tested for biological control were not simultaneously tested for tolerance to herbicides (Trumble and Kok 1982) but it appears that 2,4-D can be applied at low rates in conjunction with the rust Puccinia punctiformis to achieve better control than either treatment alone (Haggar et al. 1986).

Leaf-feeding Painted Lady Butterfly (*Vanessa cardui*)
Larvae of the native Painted Lady Butterfly (Vanessa cardui) feed on Cirsium arvense and other Cirsium species, and can defoliate and kill individual plants (Detmers 1927, Rees 1991). Painted lady typically occurs in southern states, including California, and is itself infected by a virus that keeps its populations low. Every 8-11 years, populations explode and the butterflies migrate north where they can temporarily be very effective biocontrol agents. Viral infection spreads rapidly in large painted lady butterfly populations, however, and within a year or two the butterfly populations drop again (Rees 1991).

Leaf-feeding Beetle *Cassida rubignosa*
Cassida rubignosa was accidentally introduced to the US in 1902 (Barber 1916 in Ang et al. 1995). This beetle causes severe defoliation of Canada thistle in Virginia and Maryland (Ang et al. 1995) but only minimal damage in Quebec (Forsyth and Watson 1985a). Defoliation by Cassida rubignosa is most effective at high insect density on young plants (Forsyth and Watson 1985a), but under field conditions this insect is not synchronized with young thistles and thus causes minimal damage. Ang et al. (1995) determined that Cassida rubignosa significantly reduced thistle biomass and survival. At a density of 20 beetles/plant, over two-thirds of the thistles died by the end of the growing season. Cassida impact was substantially greater during drought conditions, and roots were "devastated" by attacks of 10 beetles/plant (Ang et al. 1995). However, damage by Cassida rubignosa is rarely sufficient to reduce thistle growth in the field (Diamond 1993). Cassida rubignosa larvae are themselves parasitized by fly, wasp, and beetle species (Tipping 1993). In addition, Cassida have low dispersal rates and rarely move more than 2 m from

the release site within 26 days (Tipping 1993). Adults oviposit at the release point, regardless of thistle density (Tipping 1993). For effective control, beetles must be deposited on thistle rosettes at approximately 4 m intervals, or at least in each patch within a site.

Stem-mining weevil *Ceutorhynchus litura*
Between its initial introduction in North America in 1967 and 1985, the stem-mining weevil Ceutorhynchus litura became established in five Canadian provinces and Montana (Peschken and Wilkinson 1981, Story et al. 1985). Ceutorhynchus litura can reduce overwinter survival of Cirsium arvense but thistle stands recover by shoot recruitment from unattacked plants (Rees 1990). Females feed on leaf tissues and lay eggs in feeding cavities. The developing larvae mine leaves and migrate inside stems to the root collars (Rees 1991). Unfortunately, Ceutorhynchus litura larvae mine the parenchyma tissue of the stem pith and do not damage vascular bundles, so water translocation is not affected (Peschken and Wilkinson 1981). While thistles usually survive the stem mining, the holes left by departing larvae provide entrance sites for other arthropods, nematodes, and disease organisms which cause high mortality of belowground shoots (Rees 1990). However, production of new shoots from underground roots the following spring offsets shoot mortality caused directly and indirectly by Ceutorhynchus litura (Rees 1990).

Seed Head Predator *Orellia ruficauda*
Orellia ruficauda is a small fly that deposits its eggs in Cirsium arvense flower heads. Damage occurs when developing larvae eat the seeds in mid-summer (Detmers 1927). This may reduce seed production and seed dispersal (Forsyth and Watson 1985b). In one study 20-85% of seed heads were attacked, and 20-80% of seeds within each attacked seedhead were damaged. Forsyth and Watson (1985a) reported that Orellia ruficauda occurred in up to 70% of flowerheads and destroyed 22% (range 0-90%) of the seeds/head. Seed predation causes only limited suppression of Cirsium arvense, however, as the plant spreads primarily by vegetative means (Diamond 1993, Forsyth and Watson 1985a). While flies avoid laying eggs in male flowerheads and preferentially select female flower heads, the developing larvae do not eat enough seeds in a flowerhead to affect either the individual seed head or the population (Lalonde and Roitberg 1992b). Apparently, flies lay only a few eggs in any one flower head, and avoid laying eggs in previously-infested flower heads (Lalonde and Roitberg 1992a).

Other Natural Enemies

The Chrysomelid beetle Altica carduorum weakens Canada thistle by defoliating it and feeding on its flower heads. It was first regarded as a promising control agent because of its specificity and continuous feeding habit, but has proven unsatisfactory because of its own susceptibility to predation (Peschken et al. 1970, Story et al. 1985, Schaber et al. 1975). Cleonus piger is a root-feeding weevil that can cause wilting and plant death, but plants usually regenerate from damaged vascular tissue (Forsyth and Watson 1985a). The leaf spot disease Septoria cirsii is host specific to Cirsium arvense, and causes severe damage to Cirsium arvense plants in the field, inhibiting seed germination and root elongation, and causing leaf chlorosis and necrosis (Hershenhorn et al. 1993). This disease has been proposed for consideration as a biological control organism.

**USDA Forest Service - Biological Control of Invasive Plants in the Eastern United States - http://www.invasive.org/eastern/biocontrol/17CanadaThistle.html - Publication FHTET-2002-04 - Authors: R. Van Driesche, et al., 2002.**

There has been little work done to evaluate the effectiveness of biological control agents for Canada thistle in the eastern United States. Using Canada thistle root cuttings transplanted to caged field plots, Ang et al. (1995) showed that feeding by C. rubiginosa significantly reduced biomass and survival of Canada thistle. The effects of C. rubiginosa were stronger than those of plant competition from tall fescue (Festuca arundinacea Schreb.) and crownvetch (Coronilla varia L.). Similar results were obtained by Bacher and Schwab (2000) in Switzerland. Forsyth and Watson (1986) evaluated the stress inflicted on Canada thistle by four insect species and one pathogen in Québec, Canada. The seed head predator O. ruficauda reduced seed production by about 22%. Root mining by C. pigra sometimes killed plants. Main shoot galling by U. cardui reduced plant height and number of flowers, but side-shoot galling had less impact. Reports of the impact of C. litura have been varied. Based on field sampling, Rees (1990) suggested that this species had a significant impact on survival of Canada thistle in Montana. Peschken and Derby (1992), however, found in controlled experiments that combined attack by this species and U. cardui had no significant effect on most performance parameters of Canada thistle. The impact of biological control of Canada thistle in terms of economic benefits or recovery of native plant communities has not been evaluated.

## CHEMICAL CONTROL*:

**Illinois Nature Preserves Commission - Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/cthistle.html - Vol.1, No. 7 - Author: Max Hutchison - February 6, 1990.**

Spot application of the amine formulation of 2,4-D according to label instructions can control this plant. Individual plants of Canada thistle should be treated with a wick applicator or hand sprayer. The herbicide 2,4-D amine is selective for broadleaf plants. To reduce vapor drift, use an amine formulation of 2,4-D rather than an ester formulation. Precautions should be taken to avoid contacting nontarget plants with the solution. **Do not spray so heavily that herbicide drips off the target species**. The herbicide should be applied while backing away from the areas to avoid walking through the wet herbicide. By law, herbicides may only be applied as per label instructions and by licensed herbicide applicators or operators when working on public properties.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Canada thistle - http://www.nps.gov/plants/alien/fact/ciar1.htm - Authors: Gwendolyn Thunhorst and Jil M. Swearingen - March, 2005.**

In natural areas where Canada thistle is interspersed with desirable native plants, targeted application of a systemic herbicide such as glyphosate (e.g., Roundup or Rodeo), which carries plant toxins to the roots, may be effective. For extensive infestations in disturbed areas with little desirable vegetation, broad application of this type herbicide may be the most effective method. Repeated applications are usually necessary due to the long life of seeds stored in the soil.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Cirsium arvense - http://tncweeds.ucdavis.edu/esadocs/cirsarve.html - Author: Victoria Nuzzo - June 1997.**

Most studies of herbicide use are focused on reducing Cirsium arvense in agricultural areas, and are not directly applicable to use in natural

areas. For example, Jaeger (pers. comm.) stated that boom spray application of 2,4-D for Canada thistle control in Kilen Woods State Park, Minnesota was ineffective because it set back the succession of natural communities, actually opening areas for thistle invasion. Other herbicides can have similar impacts on native vegetation.

The following factors should be considered when using herbicides against Canada thistle:

- Their effectiveness is contingent upon Cirsium arvense growth stage, environment (Tworkoski 1992), ecotype (Hodgson 1970), and genotype (Frank and Tworkoski 1994).
- Different ecotypes respond differently to the same herbicide, so what is effective at one locale, or on one clone, may not be effective in other locales or clones (Frank and Tworkoski 1994). It is important to vary herbicides used at a site to prevent clones tolerant to one herbicide from becoming dominant (Frank and Tworkoski 1994). When selecting an alternative herbicide it is best to use one with a different mode of action (mechanism by which it kills plants) to minimize chances that plants are not tolerant to both herbicides.
- In many habitats Cirsium arvense goes dormant shortly after native species, so there is only a limited window to apply herbicides when native species will not be affected.
- It is important to treat an entire clone, as not all shoots and roots in a clone remain physically connected (Donald 1992).
- For all herbicides except 2,4-D, two or more applications give better control than a single application, regardless of seasonal sequence (spring-fall treatment gave equal control to fall-spring treatment; Zimdahl and Foster 1993, Donald 1993).
- Cirsium arvense is best treated with herbicides in early spring or in fall and fall treatments are usually more effective than spring treatments (Haderlie et al. 1987, Darwent et al. 1994a, Lym and Zollinger 1995). Herbicide absorption is enhanced in late summer and fall, when plants are in the rosette stage (Hunter et al. 1990), as shoot-to-root translocation is greatest at that time (Darwent et al. 1994a). Hunter (1996) found that control is improved if thistles are cut in late July and the resprouts treated with glyphosate about 4 weeks later in late August (the 'August rosette stage'). Second best treatment time is at flower-bud stage, when root reserves are lowest, particularly under droughty conditions (Haderlie et al. 1987). However, native species can be damaged by growing season herbicide application.

Canada thistle's deep, well-developed root systems make it resilient to most control methods including herbicides. However, Cirsium arvense undergoes several growth stages during the growing season and during certain stages root carbohydrates are depleted. Root carbohydrate depletion is related to growth stage and is greatest when flowering occurs, but replenishment is related only to environmental conditions, and generally occurs in late summer and fall. Younger growth stages (spring) are likely more susceptible to herbicide, but the root system is larger and more difficult to kill in spring before the flower stalk emerges; older growth stages (fall) are somewhat less susceptible, but the root system is depleted and smaller, and assimilates are naturally moving from the leaf tissues to the root system (Tworkoski 1992). More assimilate (and hence herbicide) moves into the roots under short days and low temperatures (fall) than long days and warm temperatures (summer; McAllister 1982).

Herbicide effect is enhanced when 1) Cirsium arvense roots are weakened during the growing season by herbicide treatment, crop competition, or frequent mowing or tilling; and 2) new shoots are stimulated to grow. Suitable herbicides (e.g. glyphosate) should be applied to new growth when leaves are green (September or October). Avoid applying herbicide to old leaves (thick cuticle limits absorption) or to drought-stressed leaves.

Clopyralid (Stinger or Transline), Clopyralid plus 2,4-D (Curtail)
Clopyralid plus 2,4-D (sold under the tradename Curtail) provides the best and most consistent control of Canada thistle in agricultural areas (Lym and Zollinger 1995), but may damage native forbs and shrubs. Clopyralid is a relatively selective post-emergence herbicide that kills certain broadleaf weeds and woody plants but does little harm to others such as members of the mustard family (Brassicaceae) or to grasses and other monocots. It is especially effective against members of the sunflower, buckwheat and pea families (Asteraceae, Polygonaceae and Fabaceae, respectively). The basis of this selectivity is not well understood for clopyralid or other auxin-type herbicides like 2,4-D or triclopyr (sold under the tradename Garlon). Clopyralid may have limited soil residual and is most effective on short (young) thistle shoots. Control was "excellent" on 5-15 cm tall shoots, very good on 30 cm tall flowering shoots; and poor on 80 cm tall shoots (Donald 1992). Annual applications in early June at 70+280 g ai/acre (clopyralid + 2,4-D) resulted in elimination or near-elimination of all Canada thistle roots in the top 50 cm of soil after 2-4 years.

Fall application of Clopyralid delayed shoot emergence by two weeks, and reduced shoot density the following summer (Donald 1993). The impact of clopyralid increased with increased application rate, and application of 840 g/ha had the greatest impact. One fall application with clopyralid at 560 g/ha prevented almost all Cirsium arvense shoot emergence the following spring.

Glyphosate (Roundup, Rodeo)
Glyphosate is a non-selective systemic herbicide that kills all vegetation green at the time of application. It has little or no soil residual. Glyphosate impacts Cirsium arvense by reducing the number of root buds and regrowth of secondary shoots, more than by reducing root biomass (Carlson and Donald 1988). No root bud regrowth occurred when glyphosate was applied at 0.28 kg/ha (Carlson and Donald 1988). Translocation of glyphosate is significantly greater in plants at the bud to flowering stage than in younger plants (Sprankle et al. 1975), and is greatest when plants are in the 'August rosette stage' (Darwent et al. 1994, Hunter 1995). The root is larger at the rosette stage, diluting the effect of glyphosate, but herbicide concentrations in the root are still up to 3 times higher at this time (Hunter 1995) because more herbicide is translocated when leaves are in vegetative (rosette and shoot-elongation) stage than in flowerbud or flowering stage (Hoefer 1982). In the laboratory, 4 times more glyphosate was translocated to the roots of rosettes than flowering plants (Hunter 1995).

Fall is the best season for applying glyphosate (Darwent et al. 1994, Lym and Zollinger 1995). For optimal results apply glyphosate under warm conditions prior to the first killing frost and when soil moisture is good, or after plants have adjusted to colder weather. Avoid treating thistles immediately before the first frost (Lym and Zollinger 1995). Plants treated with glyphosate one day before frost had much lower translocation of herbicide to the roots than plants in warm conditions, or plants hardened to cool air and soil temperatures (15/5o C; Hoefer 1982).

Response of Cirsium arvense to glyphosate varies among clones (Frank and Tworkoski 1995, Darwent et al. 1994a). The majority of damage occurs after 3 days, but glyphosate continues to act on sensitive tissues for up to 45 days (Carlson and Donald 1988). Good soil moisture is important for glyphosate to be effective (Haderlie et al. 1987). Glyphosate impact was slightly reduced under severe drought conditions (Lauridson et al. 1983).

A low glyphosate concentration (2.5%) was more effective than higher concentrations (5%, 10% and 30%) reducing shoot growth and regrowth

76% at the lower rate and having no effect at the higher rates (Boerboom and Wyse 1988b). At high concentrations glyphosate kills leaves so quickly that they are unable to translocate the herbicide to the roots before they die. Droplet size is also a factor, as large droplets kill leaf tissue more than small droplets (Boerboom and Wyse 1988b). Lower levels of surfactant (MON 0818) are recommended, as glyphosate mixed with high MON 0818 concentrations may kill leaves rapidly (Boerboom and Wyse 1988b).

Haderlie et al. (1987) stated that glyphosate was most effective on fall regrowth, then at flower/bud stage, and least effective in spring when applied to 25 cm tall plants. On the other hand, Devine (1981) found that although glyphosate translocation was slower under low temperatures, total uptake was not affected by growing conditions, and after 5 days 63% of the amount applied was absorbed and 22% exported to the roots regardless of temperature. Glyphosate was unevenly distributed in the root system and concentrated in fibrous roots and new shoot buds (Devine 1981). Between 1% and 2% of glyphosate was extruded by roots (pumped out into the surrounding soil) after 10 days (Devine 1981).

Cirsium arvense response to glyphosate differs between sites and/or clones. In Canada, a single application of glyphosate at 0.45 kg/ha reduced thistle shoot density by >75% at two sites, while 1.8 kg/ha was required to achieve the same level of control at a third site (Darwent et al. 1994). Four consecutive annual applications at 0.45 kg/ha reduced Cirsium arvense shoot density > 98%, at two sites, but at the third site four annual applications at 1.8 kg/ha were required (Darwent et al. 1994a). Most reduction occurred after the first application at all sites.

One or two applications of glyphosate at 1.7kg/ha did not prevent Cirsium arvense shoot regrowth, as enough roots remained to allow the plants to survive and resprout (Donald 1993). Fall treatment in two consecutive years decreased shoot density 94% the following fall, and root weight 77% (Carlson 1987). Five years after the treatments, however, thistle densities were the same in treated and untreated areas (Donald and Prato 1992).

Jaeger (pers. comm.) found application of Roundup to individual plants with a Walk-a-Wick applicator was difficult because the thistles were often below grass level. In 1985, park personnel in Minnesota began using a 4-5 gallon Solo backpack tank with the nozzle modified by a brass adjustment to apply a straight stream (not mist) at low pressure. Roundup at 3-4% was mixed with a purple agricultural dye and the mixture dribbled at the top of the stem and allowed to run down the stem. Use of the dye which persisted as a marker of treated plants for up to a week cut both the time involved and amount of herbicide used in half. Plants were treated in the pre-bud stage and rounds were made weekly to assure treatment of plants that were missed earlier.

Chlorsulfuron
Chlorsulfuron is a post-emergent herbicide that primarily suppresses regrowth of Canada thistle, and secondarily reduces the number of root buds and plant weight (Peterson 1983). Addition of growth regulators (chlorflurenol and dicamba) to chlorsulfuron enhanced control of Cirsium arvense in the greenhouse, but not under field conditions (Peterson 1983). Thistle density was reduced 2-5 years after spring application of chlorsulfuron (Donald and Prato 1992)

NOT RECOMMENDED

Picloram (Tordon)
Picloram is a restricted use herbicide that may persist for up to 3 years in the soil and is not registered for use in California. It is relatively soluble and thus likely to be carried to the water table by percolating rain or irrigation water. Two to three annual fall applications of picloram at 280 g/ha gradually reduced Cirsium arvense density, and both one and three consecutive annual fall applications at 560 g/ha essentially eliminated Cirsium arvense (Donald 1993). Haderlie et al. (1987) found picloram was the most effective herbicide against Canada thistle but it killed all broadleaved vegetation in treated areas. Picloram accumulates in shoot apices (Sharma and Vanden Born 1973 cited in Donald 1990) and is applied at flower bud stage or to fall regrowth (Haderlie et al. 1987).

Dicamba (Banvel)
Dicamba has limited effectiveness on Cirsium arvense, and it persists for long periods in the soil making it unacceptable for use in most natural areas. Cirsium arvense ecotypes vary in susceptibility to dicamba (Hodgson 1970, Saidak and Marriage 1976).

Metsulfuron (Ally)
Metsulfuron is ineffective against Canada thistle. Three consecutive fall applications did not reduce Cirsium arvense "sufficiently" (Donald 1993). Metsulfuron must be applied with another broadleaf herbicide, such as 2,4-D, to suppress Canada thistle (Lym and Zollinger 1995).

2,4-D
Canada thistle ecotypes varied greatly in their susceptibility to 2,4-D (Hunter and Smith 1972) and 2,4-D's impacts on treated plants were erratic (Donald 1990) and less effective than glyphosate or dicamba (Lym and Zollinger 1995). Three consecutive fall applications of 2,4-D did not reduce Cirsium arvense "sufficiently" (Donald 1993). Effectiveness of phenoxy herbicides like 2,4-D and MCPA is greater when root carbohydrate reserves are low (Marriage 1981). In agricultural situations, a combination of 2,4-D with fertilization was effective under some circumstances. Hodgson (1968) found combining 2,4-D at .24 to 2.24 kg/ha with 33.6 kg/ha nitrogen and 112 kg/ha phosphorus resulted in better thistle control and higher yields of spring wheat than either herbicide or fertilizer alone.

Five days after application 2,4-D was evenly distributed throughout the root system (Devine 1981). 3.1% of the applied 2,4-D is extruded from the roots (Devine 1981).

Bentazon (Basagran)
Bentazon is a post-emergent contact herbicide that can control topgrowth of Canada thistle when applied to plants roughly 20 cm tall in late spring and summer (Haderlie et al. 1987). At this time, however, native vegetation is very susceptible to damage. Bentazon-induced chlorosis was evident in thistles emerging 10 months after treatment, indicating that Bentazon may be stored in roots over winter and transported back to the leaves in spring (Brewster and Stanger 1980).

Thistle density decreased >80% after single applications, applied late May through late June (Brewster and Stanger 1980). Applications in early May treatment were less effective. Repeated applications (two applications at 10-14 day intervals) of 2.2 kg/ha provided better control than single applications at higher rates (3.4 - 6.7 kg/ha; Brewster and Stanger 1980). Split applications provided better control than a single application. A total of 0.84 kg/ha resulted in 84% control with one application, and 92% with two applications (Boerboom and Wyse 1988a). A total of 1.1 kg/ha

in one or two applications reduced Cirsium arvense by 40%, and by 76% when applied in four applications (Boerboom and Wyse 1988a).

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

## OTHER CONSIDERATIONS:

**Illinois Nature Preserves Commission - Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/cthistle.html - Vol.1, No. 7 - Author: Max Hutchison - February 6, 1990.**

RECOMMENDED PRACTICES ON BUFFER AND SEVERELY DISTURBED SITES:
Control procedures recommended above for high quality natural communities are also applicable to buffer and severely disturbed sites. Additional control measures are as follows. On large sites (old fields, ditch banks, roadsides) with heavy infestations, thistles should be mowed when in full bloom, and as close to the ground as possible. Cut flower-heads should be removed to prevent scattering seeds on site. Repeated mowing may be needed for several years to obtain adequate control.
A foliar application of a 1-2% solution of Roundup (a formulation of glyphosate) applied in spring when plants are 6-10 inches (15.2 -25.4 cm) tall is an effective herbicide treatment. Individual plants should be spot-treated with a wick applicator. Roundup normally kills the entire plant, including the roots, when applied in this manner. Roundup is a nonselective herbicide and precautions should be taken to avoid contacting nontarget plants with the solution. **Do not spray so heavily that herbicide drips off the target species**. As with 2,4-D amine, Roundup should be applied while backing away from the areas to avoid walking through the wet herbicide. Roundup should not be used in high-quality natural areas during the growing season because of the possibility of harming nontarget plants.
On severely disturbed sites with heavy infestations, such as cropland or abandoned cropland, the site could be plowed and sowed to a cover crop (wheat, alfalfa, rye), if practical and desirable. The following May, the cover crop should be plowed under and desired native species should be seeded.

FAILED OR INEFFECTIVE PRACTICES:
Fire early in the growing season can increase sprouting and reproduction. Prescribed burns in late spring are effective, as discussed previously. Tillage disturbance of soil may provide ideal conditions for reinvasion and for introduction of other exotics. Grazing is not an effective control measure as the prickles prevent livestock from grazing near Canada thistle.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Poison hemlock
## *Conium maculatum* L.

**SOURCE**: The following information on this species is taken from The Nature Conservancy, Invasive Species Initiative, Element Stewardship Abstract (ESA) for *Conium maculatum* at http://tncweeds.ucdavis.edu/esadocs/conimacu.html. Modifications include additional resource information. Check the website links for future updates. The last update for this ESA is May 31, 1989.

**DESCRIPTION:** Tall biennial (sometimes perennial in favorable locations) that reproduces from seeds.



]Top 2 photos: © John M. Randall/The Nature Conservancy
Bottom 2 photos: © Barry M. Rice/The Nature Conservancy

**RANGE:** *Conium maculatum* is a native of Europe, western Asia and North Africa. It was brought to the United States from Europe as a garden plant. "Poison hemlock is common and spreading in parts of the United States and Canada, particularly on the West Coast; it is common and of some importance in New Zealand, and it also occurs in South America and the British Isles. In Australia, it occurs generally throughout the southern states but has occurred as far north as Queensland" (Parsons 1973).

**HABITAT:** *Conium maculatum* "commonly occurs in sizable stands of dense, rank growth along roadsides, field margins, ditchbanks and in low-lying waste areas. It also invades native plant communities in riparian woodlands and open flood plains of rivers and streams in southern California" (Goeden and Ricker 1982) and other regions in the state. It is common on shady or moist ground below 5000 feet, especially in

cismontane California.

**REPRODUCTION:** Life Cycle: In California, "poison hemlock reproduces only from seed, both as a biennial and winter annual, and occasionally as a short-lived perennial" (Goeden and Ricker 1982). "Seeds germinate in autumn and plants develop rapidly throughout the winter and spring. Some produce flowering stems in the first spring and die in the summer. Others remain in the vegetative stage without producing flowering stems until the second spring, thus becoming a biennial. Plants are more likely to be biennial in very moist situations. After producing seeds, the plants die in the summer ... The spread of hemlock is by seeds which can adhere to farm machinery, vehicles, agricultural produce, mud and clothing as well as being carried by water and to a limited extent wind" (Parsons 1973). "Hemlock is capable of rapid establishment after autumn rains, particularly on disturbed sites or where little vegetation exists at the start of the autumn growing season. Once it is firmly established under such conditions, hemlock can preclude most other vegetation and established pastures" (Parsons 1973).

**IMPACTS:** *Conium maculatum* can be a tenacious weed particularly in moist habitats and along streams. Poison hemlock may act as a pioneer species quickly colonizing disturbed sites and displacing natives during early successional seres. The presence of *C. maculatum* degrades habitat quality and could indicate a management problem on an ecological preserve. *Conium maculatum* is poisonous to both humans and livestock. It was probably used to poison Socrates. "Poisoning of humans has occurred after the ingestion of seeds, leaves and roots and even as a result of blowing through the hollow stems when used as whistles or pea-shooters. The seeds, however, are the most toxic part of the plant. Extracts of hemlock have been used as arrow poisons by North American Indians, and it was used medicinally for many years in treating tumors, ulcers and gout" (Parsons 1973).

**AUTHOR:** Don Pitcher

**For more information on poison hemlock, please contact:**
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA-NRCS PLANTS Database, http://plants.usda.gov/

**REFERENCES:**
Fairbairn, J. W. 1970. The alkaloids of hemlock. P.361-368 in V.H. Heyward (ed.), Biology and chemistry of the Umbelliferae.
Fairbairn, J. W. and A.A.E.R. Ali. 1968. The alkaloids of hemlock (Conium maculatum L.). III. The presence of bound forms in the plant. Phytochemistry 7:1593-1597.
Fairbairn, J. W. and S.B. Challen. 1959. The alkaloids of hemlock (Conium maculatum L.). Distribution in relation to the development of the fruit. Biochemistry Journal 72 (4): 556-61.
Fairbairn, J.W. 1968. The alkaloids of hemlock (Conium maculatum L.), IV: Isotopic studies of the bound forms of alkaloids in the plant. Phytochemistry 7:1599-1603.
Freitag, J. H. and H. P. Severin. 1945. Poison-hemlock-ringspot virus and its transmission by aphids to celery. Hilgardia 16(8) :389-410.
Frohne, D. and H. J. Pfander. 1983. A colour atlas of poisonous plants. Wolfe Science, London. 291 pp.
Goeden, R. D. and D. W. Ricker. 1982. Poison hemlock, Conium Maculatum, in southern California - an alien weed attacked by few insects. Annals of Entomological Society of America 75: 173-176.
Howell, W. E. and G. I. Mink. 1981. Viruses isolated from wild carrot and poison hemlock. Plant Diseases 65:277-279.
James, L. F. and A. E. Johnson. 1976. Some major plant toxicities of the western U.S. Journal of range management 29: 356-363.
Jepson, W. L. 1951. Manual of the flowering plants of California. University of California Press, Berkeley.
Kingsbury, J. M. 1964. Poisonous plants of the U.S. and Canada. Prentice-Hall, Inc., Englewood Cliffs, NJ. 626 pp.
Leete, E. and N. Adityachaudhury. 1967. Biosynthesis of the hemlock alkaloids. II. The conversion of -coniceine to coniine and -conhydrine. Phytochemistry 6:219-223.
Muenscher, W. C. 1975. Poisonous plants of the U.S. Collier Books, New York. 277pp.
Parsons, W. T. 1973. Noxious weeds of Victoria. Inkata Press, Ltd., Melbourne, Australia. 300 pp.
Pemberton, A. W. and R. R. Frost. 1974. Celery mosaic virus in England. Plant Pathology 23: 20-24.
Robbins, W. W. 1940. Alien plants growing without cultivation in California. California Agricultural Experiment Station Bulletin 637:1-128.
Roberts, M. F. 1971. The formation of -coniceine from 5-ketooctanal by a transaminase of Conium maculatum. Phytochemistry 10:3057-3060.
Roberts, M. F. 1974. An S-adrenasyl-1-methionin; conciine methyltransferase from Conium maculatum. Phytochemistry 13: 1847-1851.
Roberts, M. F. and Richard T. Brown. 1981. A new alkaloid from South African Conium species. Phytochemistry 20:447-449.
Roberts, M.F. 1975. Y-coniceine reductase in Conium maculatum. Phytochemistry 14: 2393-2397.
Simpson, B. H. 1975. Plant toxicities in sheep on a property with unusual weed control problems. Proceedings 28th New Zealand Weed and Pest Control Conference, pp. 92-94.
Stephens, H. A. 1980. Poisonous plants of the central U.S. The Regents Press of Kansas, Lawrence. 165 pp.
Sutubutra, T. and R. N. Campbell. 1971. Strains of celery mosaic virus from parsley and poison hemlock in california. Plant Disease Reporter 55: 328-332.
Tyler, H. A. 1975. The swallowtail butterflies of North America. Naturegraph Publishing, Healdsburg, CA. 192 pp. USDA. 1980. Plants poisonous to livestock in the western States.
USDA Agricultural Information Bulletin No. 415. 90pp. Vorobyov, N. E. 1960. On some biological properties of biennial weeds of the Danube stepp. Ukrainsky Botonichny Zhurnal 17(5) : 43-49.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Poison hemlock
# *Conium maculatum* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resource:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Conium maculatum - http://tncweeds.ucdavis.edu/esadocs/conimacu.html - Author: Don Pitcher - May 31, 1989.**

## GENERAL MANAGEMENT CONSIDERATIONS:

Mechanical or chemical removal of Conium maculatum is relatively easy (see Management section below), but complete eradication may be difficult due to reintroductions and the presence of viable seeds in the soil.

Conium maculatum requires active control measures or it can become dominant on a site, particularly disturbed areas such as roadsides.

Most of the following management information was obtained through personal communication with Jim McHenry, U.C. Davis Agricultural Extension (1985).

## MANUAL AND MECHANICAL CONTROL:

Hand Pulling or Grubbing: Hand pulling works easiest with wet soils and with small infestations. When grubbing, it is not necessary to remove the entire root system since the plant is not perennial. It is best to pull or grub out the plant prior to flowering (Parsons 1973). "Follow-up cultivation is necessary to deal with any seedlings and if possible a vigorous pasture should be established to compete with any further seedling growth" (Parsons 1973). Poison hemlock remains toxic for several years after being pulled, and it is wise not to leave the dead plants where they might be eaten by wildlife or children.

Mowing: Multiple mowings close to the ground may eventually kill Conium maculatum. "Mowing or slashing of the plants just before flowering is often effective, but sometimes new growth which requires re-treatment is produced from the base" (Parsons 1973).

## BIOCONTROL:

There are no known methods of effective biological control of Conium maculatum. The methods of using viral infection and/or phytophagous insects to control this weed need more research and experimentation.

Conium maculatum is often found infected by one or more strains of virus such as ringspot virus, carrot thin leaf virus (CTLV), alfalfa mosaic virus (AMV), and celery mosaic virus (CeMV) (Freitag and Severin 1945, Howell and Mink 1981). However, the stands of poison hemlock seem to survive in spite of viral attack. It is generally common for virus-affected plants to be more often stunted than killed. An apparent example of this phenomenon is the presence of extremely high populations of the ringspot vector, the honeysuckle aphid Rhopalosiphum conii (Dvd.), occurring on Conium.

Conium "is the only plant known to be infected by ringspot in nature. The two symptoms most useful in identifying this ringspot have been the chlorotic areas and the line patterns. They can easily be detected by the mottling of the leaves and by line and ringspot patterns. Under natural conditions the infected plants are not stunted, but often show a downward curling of the leaflets along the midrib" (Freitag and Severin 1945).

Similarly, populations of CTLV, AMV, and CeMV were isolated from Conium maculatum during a survey in southeastern Washington in 1975 and 1979. "CTLV and CeMV were the most prevalent viruses in wild carrot and poison hemlock of southeastern Washington. CTLV and CEMV were each recovered from 9% of the wild carrots and from slightly more than 20% of the poison hemlock, with 7% and 11% infected by both CTLV and CeMV, respectively ... AMV was found only at one location, infecting four poison hemlock plants" (Howell and Mink 1981).

"The incidence of CTLV and CeMV ranged from 0 to more than 80 percent. This variation appeared related to moisture availability. Where water was short through the summer, many of the second-year biennials matured and died before new plants emerged, thus decreasing the probability of virus transmission from the older to the younger weeds ... Poison hemlock, which is abundant in southeastern Washington, is considered a natural reservoir for CeMV in England (Pemberton and Frost 1974) and California (Sutabutra and Campbell 1971)" (Howell and Mink 1981).

However, as in the ringspot virus example cited above, use of these viruses as Conium maculatum controlling agents would depend upon (1) how they affect the viability of poison hemlock and (2) the feasibility of using an agent in the wild that could also adversely affect agricultural crops.

The useability of phytophagous insects to control Conium maculatum needs more experimentation. The phytophagous insect fauna of poison hemlock in southern California is largely comprised of relatively unspecialized, polyphagous, ectophagous, sap- and foliage-feeding species. Thereof, poison hemlock hosted amazingly few insect species or individuals. A clear majority, 16 of the 20 phytophagous insect species found on this weed, were rare.

"Substantial, but unquantified seed destruction by Hyadaphis foeniculi was noted at several locations, but otherwise poison hemlock suffered little insect injury. Most parts of this weed remain essentially free of deleterious insect attack. Apparently, the century since this weed was accidentally introduced into California (Robbins 1940) has provided sufficient opportunity for only very few native phytophagous insects to overcome its toxic defenses and transfer to this colonizing plant species" (Goeden and Ricker 1982).

"The larval 'anise swallowtail' usually feeds on Umbelliferae. This butterfly may be in the process of adopting poison hemlock as an additional food plant in California ... Foreign exploration for natural enemies of poison hemlock in Europe, especially in areas of Mediterranean climate for use in California, is indicated as the next step in ascertaining whether the relative trophic vacuum that this weed represents might be usefully filled by a complex of intentionally introduced, specialized natural enemies" (Goeden and Ricker 1982).

## CHEMICAL CONTROL*:

If extensive areas are covered with Conium maculatum, chemical controls are simpler and less labor intensive.

2,4-D in moderate doses does not kill grasses (except the more susceptible bentgrass). It is most effective against poison hemlock when the ester form is mixed with diesel oil to allow penetration of the leaves and stems. It can be used to hand spot (the most effective technique), or to spray larger areas. The suggested mixture is 1.5 lbs acid equivalent per acre. Mix 2 quarts of diesel oil with 1.5 lbs of 2,4-D ester and add to 100 gallons of water in a spray tank. A 100-gallon tank should cover approximately one acre.

Banvel (active ingredient Dicamba) also works on broad-leaved plants but not as effectively as 2,4-D. The suggested mixture is 1/2 to 3/4 per 100 gallons of water and a surfactant is required.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




# Crown vetch
## *Coronilla varia* L.

SOURCE: The following information on this species is taken, in part, from the Illinois Nature Preserves Commission, Illinois Vegetation Management Guidelines for Trailing Crown Vetch, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website link for future updates. The last update for this fact sheet is February, 1990.

**DESCRIPTION:** Trailing crown vetch is an herbaceous legume with creeping stems 2-6 feet (0.6-1.8 meters) long, bearing leaves consisting of 15-25 pairs of oblong leaflets. This species has strong rhizomes, and a reclining growth habit. The pinkish lavender to white clusters of flowers occur in umbels on long, extended stalks. Flowers produce narrow, finger-like pods containing slender seeds that have been reported to be poisonous.







Photos: Dan Tenaglia, www.missouriplants.com, www.forestryimages.org

**SIMILAR SPECIES:** Trailing crown vetch is distinguished from other plants in the legume family by the following characteristics: 1) compound leaves with an odd number of leaflets ranging between 15-25, 2) the presence of leaves and flower stalks arising from the main stem, and 3) the occurrence of flowers in an umbel. Trailing crown vetch should be accurately identified before attempting any control measures. If identification of the species is in doubt, the plant's identity should be confirmed by a knowledgeable individual and/or by consulting appropriate books.

**DISTRIBUTION:** The natural distribution of trailing crown vetch is Europe, southwest Asia and northern Africa. It is introduced or naturalized in the United States from Maine to South Dakota, south to Virginia, West Virginia, Kentucky, Illinois, and Missouri. This species is widely distributed in Illinois, with occurrences recorded from over one-half of the counties.

**HABITAT:** Trailing crown vetch has been grown extensively in the northern two-thirds of the United States for temporary ground cover, erosion control, and as a green fertilizer crop. It is also used as a bank stabilizer along roads and waterways. This plant prefers open, sunny areas. It occurs along roadsides and other rights-of-way and in open fields.

**LIFE HISTORY**: Flowers appear from May to August. Trailing crown vetch is a perennial, herbaceous plant that can spread rapidly by seed and its multi-branched creeping root system.

**EFFECTS UPON NATURAL AREAS:** Trailing crown vetch is a serious management threat to natural areas due to its seeding ability and rapid vegetative spreading by creeping roots. This aggressive exotic is now widespread in Illinois along roadsides and in waste grounds, from where it becomes a serious invader of prairies and dunes.

Photo: Dave Powell, USDA Forest Service, www.forestryimages.org.



**AUTHOR**: Jim Heim, Illinois Department of Conservation, Oregon, Illinois

**For more information on crown vetch, please contact:**
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Southeast Exotic Pest Plant Council; Invasive Plant Manual, http://www.se-eppc.org/manual/
The Bugwood Network, MA-EPPC Invasive Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Northeastern Area, Invasive Plants-Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program, Fact Sheets, http://www.dcr.virginia.gov/dnh/invlist.htm
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/scott/weed_id/genusindex.htm
Wisconsin Department of Natural Resources, Invasive Plant Information Sheets,
http://www.dnr.state.wi.us/org/land/er/invasive/nonnative.htm#aquatics

**REFERENCES**: Gleason, H. A. 1952. The new Britton and Brown illustrated flora of the northeastern United States and adjacent Canada. Vol 2. The New York Botanical Garden, New York. 655 pp.
Fernald, M. L. 1950. Gray's manual of botany, eighth edition. American Book Co., New York. 1632 pp.
Mohlenbrock R. H. 1986. Guide to the vascular flora of Illinois. Southern Illinois University Press, Carbondale. 507 pp.

**PERSONAL COMMUNICATIONS**: Glass, William. 1989. Division of Natural Heritage, Illinois Department of Conservation, Springfield, Illinois.
Nyboer, Randy. 1989. Division of Natural Heritage, Illinois Department of Conservation, Springfield, Illinois.
Packard, Steve. 1989. The Nature Conservancy, Chicago, Illinois.
Schwegman, John. 1989. Division of Natural Heritage, Illinois Department of Conservation, Springfield, Illinois.
West, Andy. 1989. Division of Natural Heritage, Illinois Department of Conservation, Springfield, Illinois.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Crown vetch
# *Coronilla varia* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Trailing Crown Vetch - http://www.inhs.uiuc.edu/chf/outreach/VMG/tcvetch.html - Vol.1, No. - Authors: Jim Heim - February 6, 1990.**

*Initial efforts in areas of heavy infestation*
Very little research information is currently available regarding the control of crown vetch. Research has largely been restricted to the establishment and management of the perennial legume for cultural purposes. As a result, a limited number of control measures only recently have been available from unpublished field notes of active natural resource managers. The following controls have met with some success, but field research is needed to adequately address this alien species. In fire-adapted communities, prescribed burning in late spring can be an effective control. Prescribed burns may need to be repeated for several years to achieve adequate control. Where feasible, late-spring mowing for several successive years can control this species.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

As with all prolific invaders, the key to the successful control of C. varia is to prevent new infestations or to control them as soon as possible. If controlled during the early stages of invasion, the potential for successful management is high. Since C. varia can reproduce both vegetatively and/or sexually, has a wide range of adaptability, and has few pests and predators, it becomes difficult to manage once firmly established. As such, the potential for large-scale restoration of wildlands where C. varia has become established is probably medium.

The best control of C. varia is probably with the use of an integrated management approach. The use of manual and mechanical methods, then another control treatment (such as a herbicide spray to control seedlings) repeated for several years, followed by active restoration efforts may be necessary to obtain desired results.

**Wisconsin Department of Natural Resources - Non-Native Plant Information - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/crown.htm**

Very little research information is currently available regarding the control of crown vetch. Research has largely been restricted to the establishment and management of this perennial legume. As a result, a limited number of control measures have become available from the unpublished notes of active natural resource managers. Further field research is needed to adequately address this species. However, preventative measures can and should be implemented: do not use crown vetch for erosion control. Encourage your local highway department to stop using it and replace it with less invasive species for roadside use

## MANUAL AND MECHANICAL CONTROL:

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Crown vetch - http://www.se-eppc.org/manual/COVA.html -2003**

Hand pulling of mature plants can be effective when controlling small initial infestations. Mowing plants in the flower bud stage for two or three consecutive years may reduce the vigor and control further spread. Plants should be cut before seeds mature and as low to the ground as possible. Impact to adjacent native plants should be minimized as much as possible.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

Manual and/or mechanical methods of plant removal can control C. varia in small, isolated patches. These methods however, are very time and labor-intensive, as all pieces of the stems, roots, and strong rhizomes must carefully be removed to avoid resprouting. Populations must also be monitored for several years following plant removal because seeds stored in the soil seed bank may germinate.

Cutting or mowing C. varia at a frequency of less than once per year is not effective at controlling populations, but does offer a method for temporarily stopping or slowing its spread. Heim & Glass (1989) report that mowing around the periphery of the desired vegetation (natural area) will keep C. varia from spreading into the area vegetatively, but does not prevent the arrival of seeds. Heim & Glass (1989) and Heim (1990) also report that repeated mowing (several times per year over several years) can eventually control C. varia. Repeated late spring mowing appears to be the most effective. Cutting or mowing can also remove standing biomass, so spray herbicides have higher efficacy.

**Wisconsin Department of Natural Resources - Non-Native Plant Information - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/crown.htm**

Where feasible, late spring mowing for several successive years can control this species. Another technique is to mow twice every year: in June and in late August, corresponding with successive leaf-out periods.

## GRAZING:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

C. varia is good quality forage and is highly palatable to cattle, horses, goats and sheep. Horses, however, should not ingest C. varia in large quantities (Cornell University 2001). When C. varia is properly grown and managed as a pasture crop, grazing does not negatively affect its rate of growth. If pastures are continually (or near-continually) grazed, other non-native species will probably invade these pastures (Barnes & Dempsey 1992). Grazing by itself therefore, does not appear to control C. varia.

Grazing can, however, be used in combination with a herbicide treatment for good control. Grazing works similarly to cutting or burning, in that it first removes most of the C. varia aboveground biomass, which ensures good herbicide spray coverage on its leaves and stems, resulting in good kill results.

## PRESCRIBED FIRE:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Trailing Crown Vetch - http://www.inhs.uiuc.edu/chf/outreach/VMG/tcvetch.html - Vol.1, No. - Authors: Jim Heim - February 6, 1990.**

A regular fire regime should control this species in fire-adapted communities. In Illinois, trailing crown vetch primarily invades communities that are fire-adapted.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

If C. varia is not dominant, prescribed burns in late spring can be an effective control treatment, especially if the surrounding native ecosystem is fire-adapted. Burning may need to be repeated for several years to achieve adequate control (Heim 1990).

At sites where C. varia is abundant, however, burning alone is not an effective control option. Burning is effective against seedlings or in slowing the spread of C. varia, but large populations will not be controlled because C. varia does not produce enough fuel to carry a hot fire. Heim & Glass (1989) report that in large infestations with sufficient fuel, fire will set back the periphery of the infestation, but the center of the population will be left unaffected.

**Wisconsin Department of Natural Resources - Non-Native Plant Information - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/crown.htm**

In fire-adapted communities, prescribed burning in late spring can be an effective control. Burns may need to be repeated for several years to achieve adequate control.

## BIOCONTROL:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

There are no available biocontrol agents for C. varia. Coleophora colutella (Lepidoptera: Coleophoridae) has recently been reported as immigrating into the U.S., and its larvae feed on C. varia (Hoebeke et al. 1993). Wheeler (1991) reports that Lepyronia coleoptrata (Homoptera:

Cercopidae), a spittlebug from Europe that feeds upon C. varia, has recently expanded its range into the northeastern U.S. Due to the continued use of C. varia as a forage, pasture, and erosion control plant, it is unlikely that a biocontrol agent will be introduced for its control.

## CHEMICAL CONTROL*:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Trailing Crown Vetch - http://www.inhs.uiuc.edu/chf/outreach/VMG/tcvetch.html - Vol.1, No. - Authors: Jim Heim - February 6, 1990.**

RECOMMENDED PRACTICES ON BUFFER AND SEVERELY DISTURBED SITE: Initial efforts in areas of heavy infestation - Same as given above for heavily infested areas. In addition, the following chemical control methods are effective and could be used where prescribed burning or mowing are not feasible.

The herbicide 2,4-D amine (dimethylamine salt of 2,4-D) is a low volatility formulation that can be foliar applied in early spring when crown vetch is growing actively. 2,4-D amine should be applied by hand sprayer at the recommended application rate on the label for spot application. Phenoxy herbicides are broadleaf selective plant growth regulators that will not harm grasses but precautions must be taken in the vicinity of sensitive nontarget plants. To reduce vapor drift, use an amine rather than an ester formulation of 2,4-D. The control area should be spring-or fall-burned to remove accumulated plant litter to insure complete foliar coverage. A follow-up treatment using recommended label rates is necessary to obtain complete results. The herbicide mecamine (2,4-D plus Dicamba) is effective when foliar applied as a 1% solution to crown vetch plants in a state of rapid growth (spring or early summer before flowering). In addition, Roundup (a formulation of glyphosate) is a broad spectrum, translocated herbicide that can be foliar applied as a 1 or 2% solution during early spring when the plant is actively growing. Avoid direct application to any body of water, sensitive species, or areas that need to be protected from drift and direct application. Roundup is nonselective and care should be taken to avoid nontarget plants. To insure good foliar coverage, the previous year's growth should be prescribed-burned to eliminate duff accumulation. A follow-up application of Roundup may be necessary the following fall or early spring to combat regeneration from underground parts or seeds.

When applying any herbicide described above, spot application should be done with a hand sprayer and should be uniform such that the entire leaf is wetted. **Do not spray so heavily that herbicide drips off the target species**. Care should be taken to avoid contacting non-target species. Native plants, left unharmed, will be important in recolonizing the site after crown vetch is controlled. The herbicide should be applied while backing away from treated areas so as not to walk through the wet herbicide. By law, herbicides only may be applied according to label instructions and by licensed herbicide applicators or operators when working on public properties.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Crown vetch - http://www.se-eppc.org/manual/COVA.html -2003**

Spot Treatments: For herbicidal controls to be effective, herbicides should be applied while the plants are actively growing. Glyphosate, triclopyr and metsulfuron have been shown to be effective in controlling crown vetch. A 1%-2% solution triclopyr or glyphosate thoroughly mixed with water is effective during the vegetative stage prior to branching or during flowering. Triclopyr is selective to broad-leaved plants and can be used in areas where native grasses are intermingled with the target plants. Glyphosate is non-selective and will affect any plant it comes in contact with. Metsulfuron should be applied at a rate of 0.3g/gallon of water. Treatments should cover the leaves and stems of plants to the point of runoff. The addition of a non-ionic surfactant at a concentration of 0.5% improves the effectiveness of foliar treatments. Read the herbicide label thoroughly prior to use.

Broadcast: Broadcast treatments are appropriate for large infestations such as fields or prairies. Since native plants will be intermingled with lespedeza, triclopyr and metsulfuron are the preferred herbicides due to their selective characteristics. Apply triclopyr at a rate of 1.0-1.5 pints per acre. Metsulfuron should be applied at a rate of 0.5 oz per acre. Use a non-ionic surfactant according to manufacturer's instructions to improve effectiveness.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

Herbicides are currently the most effective means to control large infestations of C. varia. Herbicides for C. varia control can be applied with either backpack sprayers (to minimize overspray) or with a boom. Complete coverage of all stems of C. varia is necessary for good control (i.e., spray to wet). Higher rates of efficacy are generally obtained if the spray herbicide treatment is preceded by the removal of the accumulated plant litter (by first burning, mowing, or grazing) to ensure good foliar coverage. Heim (1990) and Heim & Glass (1989) indicate that 2,4-D, glyphosate, triclopyr, or clopyralid may be used to control C. varia.

2,4-D amine can be foliar-applied for good control in early spring when C. varia is actively growing. It will generally kill the aboveground portions of the plant(s), but some plants may survive (Heim & Glass 1989). If spot applying by a hand sprayer, Heim (1990) suggests using it at the recommended label rate. He also recommends using the 2,4-D amine formulation instead of the ester formulation, as it has a lower level of volatility and does not drift as readily. 2,4-D is a dicot-specific herbicide, and should not harm grasses or other monocots. Follow-up treatments are usually necessary to obtain good control results. A combination of 2,4-D with dicamba (tradename Weed-b-Gonâ) can also be successful (Harper-Lore 1996).

Glyphosate (tradename RoundUpâ) can be foliar applied as a 1% or 2% solution, during early spring when the plant is actively growing. Heim (1990) adds that follow-up treatments are necessary the following fall or early spring.

Triclopyr (tradename Garlon 3Aâ) is also effective at controlling C. varia. Heim & Glass (1989) report that a 2% solution of triclopyr kills 99% of C. varia in large infestations.

Clopyralid (tradename Translineâ) is a more target-specific herbicide than glyphosate, 2,4-D, or triclopyr, controlling only certain families of dicot plants, including the legumes (Fabaceae), composites (Asteraceae) and smartweeds (Polygonaceae). Bryon Walters (pers. comm.) of the Illinois Natural Areas Improvements reports that a 0.25% solution of clopyralid with 0.5% surfactant can kill up to 100% of C. varia cover (see "Examples" section below for more details).

C. varia is tolerant to both imazethapyr and imazapic (tradename Plateauâ) herbicides. Beran et al. (1999) and Masters et al. (1996) both determined when conducting experiments on grassland restoration, that C. varia is not only tolerant to these herbicides, but that its rate of establishment was actually improved after its competitors (namely Cirsium arvense-Canada thistle) were treated and reduced in abundance with these compounds.

**Wisconsin Department of Natural Resources - Non-Native Plant Information - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/crown.htm**

The herbicide 2,4-D amine (dimethylamine salt of 2,4--D) is a low volatility formulation that can be foliar-applied in early spring when crown vetch is growing actively. 2,4-D amine should be applied by hand sprayer at the recommended application rate on the label for spot application. Phenoxy herbicides are broadleaf-selective plant growth regulators that will not harm grasses, but precautions must be taken in the vicinity of non-target broad-leaved plants. To reduce vapor drift, use an amine rather than an ester formulation of 2,4-D.

A 2% active ingredient (a.i.) solution of triclopyr in water has also been successful in controlling large infestations. Like 2,4-D, triclopyr is advantageous because it is dicot-specific and does not affect grasses beyond some temporary browning.

Glyphosate is a broad-spectrum, translocated herbicide that can be foliar-applied as a 1 or 2% a.i. solution during early spring when the plant is actively growing. Glyphosate is nonselective, and care should be taken to avoid non-target plants. To insure good foliar coverage, the previous year's growth should be burned to eliminate duff accumulation and to expose new growth. A follow-up application of glyphosate may be necessary the following fall or early spring to combat regeneration from underground parts or seed.

The herbicide clopyralid has been used successfully to treat roadside populations of crown vetch. This herbicide shows promise because it is even more specific than triclopyr in the plant families it affects; specifically, this herbicide kills leguminous species but does not affect grasses and most other plant families. However, further research is needed.

When applying any of the herbicides described above, spot applications should be done uniformly with a hand sprayer to ensure that the entire leaf is wetted. Do not spray so heavily that herbicide drips off the target species. Native plants, left unharmed, will be important in recolonizing the site after crown vetch is controlled. Reseeding of native plants may be necessary where infestations of crown vetch are severe. Planting an intermediate cover crop may be appropriate.

All of these methods may need repeated applications over several years to effectively eradicate populations of crown vetch that are well established.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Coronilla varia - http://tncweeds.ucdavis.edu/esadocs/corovari.html - Author: Mandy Tu - January 2003.**

Restoration/Competition:
C. varia is a strong competitor and is generally able to outcompete and suppress native vegetation once it has invaded. If C. varia populations are reduced, however (by herbicide, repeated mowing, etc.), the native plants (if still present) are usually able to rapidly recolonize sites (S. Moats, pers. comm.; Heim 1990). Art Gover (pers. comm.) suggests selecting a variety of cool-season native grasses for competition. He recommends the use of switchgrass (Panicum virgatum) where it is native, because it is taller than C. varia and may shade it.

In studies of Canada thistle (Cirsium arvense) control -a non-native to North America despite its common name- C. varia was able to successfully compete with it under certain conditions. In wet years, C. varia suppresses Canada thistle better than defoliation alone (Ang et al. 1994a,b). Drought reduces the competitiveness of C. varia, resulting in higher densities of thistles the following year (Tipping 2001).

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Hairy willow-herb
## *Epilobium hirsutum* L.

**SOURCE:** The following information on this species is taken from the Invasive Plant Atlas of New England (IPANE) website at http://invasives.eeb.uconn.edu/ipane/. Modifications include additional resource information. Check the website link for future updates.

**DESCRIPTION:** *Epilobium hirsutum* is an herbaceous perennial that spreads by rhizomes. The stems are erect and softly hirsute-pubescent, measuring 0.5-2 m (1.5-6.5 ft.) in height. The leaves are opposite and lanceolate to oblong-lanceolate in shape. They are sessile, often clasping at the base. The leaves, growing up to 5-12 cm (2-4.75 in.) long by 1-3 cm (0.4-1 in.) wide, are sharply serrulate on their margins. These leaves are pubescent on both the upper and the lower surfaces.

The rose-colored flowers are about 2.5 cm (1 in.) across and have four notched petals. The flowers are axially arranged. The petals are pubescent at their base. The fruit are 5-8 cm long (2-3 in.). The seeds are oblong and flattened, measuring 1 mm (0.04 in.) long and have a tuft of long, nearly white hairs.

Page References Crow & Hellquist 210, Fernald 1058, Gleason & Cronquist 316, Holmgren 297, Magee & Ahles 767, Newcomb 160, Peterson & McKenny 270. See reference section below for full citations.




USDA-NRCS PLANTS Database / Britton, N.L., and A. Brown. 1913. *Illustrated flora of the northern states and Canada*. Vol. 2: 591.

**SIMILAR SPECIES**: *Epilobium angustifolium* L. (Fireweed, great willow-herb). *E. angustifolium* differs from *E. hirsutum* in that its leaves are longer, measuring 15-20 cm (6-8 in.), and that it does not have pubescent leaves.

**REPRODUCTIVE/DISPERSAL MECHANISMS**: Epilobium hirsutum spreads vigorously by way of rhizomes. It can also spread by wind-dispersed seeds, though this method is secondary to its vegetative reproduction.

**DISTRIBUTION**: This plant is a native of Eurasia and North Africa, though it is now found in Europe, Africa, Asia, southern Australia (as of 1990) and the U.S. In the United States it is found from Maine to Wisconsin and South from Illinois to Maryland. It is also found in the state of Washington. It has been reported from all the states of New England.

**HISTORY OF INTRODUCTION IN NEW ENGLAND**: The earliest record of *Epilobium hirsutum* in the Northeast is from Newport, Rhode Island in 1829. After this date there is a gap until the 1850s and 1860s when it was reported in a number of sites along the northeastern seaboard (Stuckey, 1970). The records from the 50s and 60s suggest that it may have been brought to the U.S. in ship ballast. It may have been purposefully cultivated, but soon after it became noted as a garden weed. After its arrival in coastal areas it spread inland at a fairly rapid pace.

**HABITATS (IN NEW ENGLAND):** Edge, Herbaceous Wetland, Lake or Pond, River or Stream, Wet Meadow, Yard or Garden

*Epilobium hirsutum* grows in moist to wet soils and is generally found in wetland to semi-aquatic habitats. It has the ability to adapt to wet conditions through the development of arenchymal tissues in its stem and rhizomes. It prefers open conditions where it can receive ample light. Its habitat preferences and pattern of spread are similar to that of *Lythrum salicaria*.

**THREATS:** *Epilobium hirsutum* can form dense monotypic stands in natural wetland areas capable of crowding out native species.

**For more information on hairy willow-herb, contact:**
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
USDA - NRCS PLANTS Database, http://plants.usda.gov/

**REFERENCES:**
Barakat, H.H., S.A.M. Hussein, M.S. Marzouk, I. Merfort, M. Linscheid, M.A.M. Nawwar. 1997. Polyphenolic metabolites of Epilobium hirsutum. Phytochemistry 46 (5): 935-941.
Britton, N. L. and A. Brown. 1970. An Illustrated Flora of the Northeastern United States vol. 2. Dover Publications Inc., New York.
Crow G.E. and C.B. Hellquist. 2000. Aquatic and Wetland Plants of Northeastern North America. Vol #1. University of Wisconsin Press, Madison
Fernald, M.L. 1950. Gray's Manual of Botany 8th ed. American Book Co., Boston.
Gleason H.A. and A.C. Cronquist. 1991. Manual of Vascular Plants of the Northeastern United States and Adjacent Canada. 2nd ed. New York Botanical Garden, Bronx, New York.
Holm, L.G., Pancho, J.V., Herberger, J.P., and Plucknett, D.L. (1979). A Geographical Atlas of World Weeds. John Wiley and Sons, New York, USA.
Holmgren N.H. 1998. Illustrated Companion to Gleason and Cronquist's Manual. New York Botanical Garden, Bronx, New York.
Magee D.W and H.E. Ahles. 1999. Flora of the Northeast. University of Massachusetts Press, Amherst.
Newcomb N. 1977. Newcomb's Wildflower Guide. Little Brown, Boston.
Peterson R.T. and M. McKenny. 1968. A field Guide to Wildflowers of Northeastern and North-central North America. Houghton Mifflin, Boston.
Stuckey, R.L. 1970. Distributional History of Epilobium hirsutum in North America. Rhodora 72: 164-181.
USDA, NRCS. 2001. The PLANTS Database, Version 3.1. (http://plants.usda.gov). National Plant Data Center, Baton Rouge, LA 70874-4490 USA.

**Management and Control Information**

Return to List




## Species Management and Control Information

### Hairy willow-herb
### *Epilobium hirsutus* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

**Management information for this species was unavailable at time of tutorial development**

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Tall fescue
## *Festuca elatior* L.

**SOURCE:** The following information on this species is taken from James H. Miller. 2003. Nonnative invasive plants of southern forests: a field guide for identification and control and the Virginia Department of Conservation and Recreation and the Virginia Native Plant Society, Invasive Alien Plant Species of Virginia Fact Sheets, http://www.dcr.virginia.gov/dnh/invlist.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is April, 2004.

**Synonyms**: *Festuca arundinacea* Shreb., *Lolium arundinaceum* (Schreb.) S.J. Darbyshire, meadow fescue, Kentucky 31 fescue
**Plant.** Erect, tufted cool-season perennial grass 2 to 4 feet (60 to 120 cm) in height, green in winter and spring, during which it is the most common green bunchgrass. Dark-green leaves appearing in late winter, usually flowering in spring (infrequently in late summer). Semidormant during heat of summer, with whitish seedstalks persisting. Growth resuming in fall and continuing into early winter.
**Stem**. Moderately stout, unbranched, hairless with round cross section and one to three swollen light-green nodes widely spaced near the base.
**Leaves.** Mostly basal and a few alternate, flat and long-lanceolate, 4 to 18 inches (10 to 45 cm) long and 0.1 to 0.3 inch (3 to 8 mm) wide. Whitish to yellow-green flared collars, with collar backs often at an angle to the stem. Blades smooth to rough, with one to two leaves along the stem becoming smaller upward. Midvein not apparent. Ligule a tiny white membrane.
**Flowers**. March to June (to October). Loosely branched terminal panicles, 4 to 12 inches (10 to 30 cm) long, that are erect or nodding at tips, narrow then spreading in spring, and then narrow again in summer. Spindle-shaped clusters along branches. Branches shorter upward, with four to seven flowers per branch. Flowers greenish white and shiny becoming purplish. Spikelets hairless, ellipsoid with a pointed tip.
**Seeds.** May (to November). Husked grain, spindle-shaped, 0.1 to 0.2 inch (3 to 5 mm) long. Whitish straw-colored husks, usually tipped with a short hair.




Photos: James H. Miller, USDA Forest Service, www.forestryimages.org.







Photos: Ted Bodner, Southern Weed Science Society,




www.forestryimages.org.

**Habitat:** Tall fescue can be found in disturbed areas including pastures, abandoned fields, roadsides, and railroad embankments. Tolerant of a wide range of moisture conditions, it has been planted for erosion control along levees and stream banks. It grows well under a variety of soil conditions, including nutrient-poor, acid soils.

**Distribution**: Introduced from Europe in the late 1800s, tall fescue is found throughout the United States and southern Canada. It is often grown for pasture or hay, and is spread further through the manure of grazing animals. Marketed as Kentucky 31, it is included in many lawn seed mixtures because it is easily established and drought resistant. It is found throughout Virginia.

**Threats**: As the density of tall fescue increases at a site, the diversity of native species declines. This is partly due to a natural toxin tall fescue produces to inhibit the growth of competing species. In natural areas tall fescue crowds out other species by forming dense clumps with a thick mat of roots.

**Alternatives to tall fescue:** Since the discovery of an endophyte fungus toxic to livestock which is associated with this species, many native species of warm season grasses have become popular and highly beneficial agricultural alternatives. These species are better able to tolerate drought as well as being more productive than fescue. Big bluestem (*Andropogon gerardii*), little bluestem (*Schizachyrium scoparium*), Indian grass (*Sorghastrum nutans*), switchgrass (*Panicum virgatum*), sideoats grama (*Bouteloua curtipendula)*, and Eastern gamagrass (*Tripsacum dactyloides*) have all been successfully used where fescue once was the favorite forage. More information on native warm season grasses can be obtained from the Virginia Department of Game and Inland Fisheries, P.O. Box 11104, Richmond, VA, 23230-11041. There are several fine-bladed fescues superior to Kentucky 31 for use in lawns. One is Chewning's fescue, four varieties of which are Jamestown 2, Longfellow, Shadow E, and SR 500. Another is creeping red fescue, two varieties of whichare Flyer and Shademaster. For more information contact the Virginia Cooperative Extension Service in your area.

**For more information on tall fescue, please contact:**
Illinois Nature Preserves Commission, Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild Fact Sheets, http://www.nps.gov/plants/alien/fact/pocu1.htm
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Nonnative Invasive Plants of Southern Forests, http://www.invasive.org/eastern/srs/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program Fact Sheet, http://www.dcr.virginia.gov/dnh/invlist.htm

**Management and Control Information**

Return to List




## Species Management and Control Information

# Tall fescue
# *Festuca elatior* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## MANUAL AND MECHANICAL CONTROL:

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Alien Plant Species in Virginia - Fact Sheets - Tall Fescue - http://www.dcr.virginia.gov/dnh/invlist.htm**

Mechanical methods are virtually useless in controlling tall fescue because of the thick root system and vegetative resprouting.

## PRESCRIBED FIRE:

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Alien Plant Species in Virginia - Fact Sheets - Tall Fescue - http://www.dcr.virginia.gov/dnh/invlist.htm**

The most effective method for severe infestations is burning early in the growing season, followed by spot application late in the growing season of a glyphosate herbicide to plants missed by the fire. Light infestations may be controlled by spot application of a glyphosate herbicide. These methods are ineffective if the plant is in a dormant phase. Glyphosate herbicides are recommended because they are biodegradable. However, glyphosate is a nonselective, systemic herbicide that affects all green vegetation. To be safe and effective, herbicide use requires careful knowledge of the chemicals, appropriate concentrations, and the effective method and timing of their application. Consult an agricultural extension agent or a natural resource expert for more information.

## CHEMICAL CONTROL*:

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Alien Plant Species in Virginia - Fact Sheets - Tall Fescue - http://www.dcr.virginia.gov/dnh/invlist.htm**

The most effective method for severe infestations is burning early in the growing season, followed by spot application late in the growing season of a glyphosate herbicide to plants missed by the fire. Light infestations may be controlled by spot application of a glyphosate herbicide. These methods are ineffective if the plant is in a dormant phase. Glyphosate herbicides are recommended because they are biodegradable. However, glyphosate is a nonselective, systemic herbicide that affects all green vegetation. To be safe and effective, herbicide use requires careful knowledge of the chemicals, appropriate concentrations, and the effective method and timing of their application. Consult an agricultural extension agent or a natural resource expert for more information.

**USDS Forest Service - Nonnative Invasive Plants of Southern Forests: a field guide for identification and control - http://www.invasive.org/eastern/srs/GM.html - Gen. Tech. Rep. SRS-62. Asheville, NC - Author: James H. Miller - 2003.**

Recommended control procedures:

- On forest lands, apply a glyphosate herbicide as a 0.5-percent solution in water (2 quarts per 10 gallons mix per acre), or Arsenal AC** as

a 1-percent solution (25 ounces per 20 gallons mix per acre) in spring.

- On noncroplands, apply 10 to 12 dry ounces of Plateau per 20 gallons mix per acre (consult the label for additives) in spring. Mixing Plateau with a glyphosate herbicide or Arsenal AC** will improve control but may damage associated native plants. Vantage (sethoxydim), Poast (sethoxydim), Assure (quizalofop), and Select (clethodim) may be useful on pastures, but they are usually more costly than a glyphosate mix with Plateau or Arsenal AC**.
- Early spring burning-if repeated-inhibits fescue and encourages native warm-season grasses.

** Nontarget plants may be killed or injured by root uptake.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# English Ivy
# Hedera helix L.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Some pictures have been added from www.invasive.org. Specific species management and control information is provided elsewhere. Check the website links provided to check for future updates. The last update for this PCA fact sheet is December 2000.

**NATIVE RANGE**: Europe, western Asia, and northern Africa

**DESCRIPTION**: English ivy is an evergreen climbing vine in the ginseng family (Araliaceae). Vines attach to the bark of trees, brickwork, and other surfaces by way of numerous, small root-like structures, which exude a glue-like substance. Older vines are known to reach a foot in diameter.



Photo: Jil M. Swearingen, U.S. National Park Service, Washington, DC

Leaves are dark green, waxy, somewhat leathery, and are arranged alternately along the stem. English ivy has many recognized leaf forms, the most common being a 3-lobed leaf with a heart-shaped base. Leaves in full sun are often unlobed, oval and have wedge-shaped bases.



Photo: James H. Miller, USDA Forest Service, www.invasive.org.

Umbrella-like clusters of small, greenish-white flowers appear in the fall if sufficient sunlight is available. Fruits mature in Spring and are black with a fleshy outer covering enclosing one to a few hard, stone-like seeds.

NOTE: Compounds in English ivy are somewhat toxic and include glycosides that cause vomiting, diarrhea, nervous conditions and dermatitis in sensitive individuals. This characteristic helps ensure spread of the seeds by many native songbirds that are attracted to the black berries in Spring when other food sources are limited.

**ECOLOGICAL THREAT**: English ivy is an aggressive invader that threatens all vegetation levels of forested and open areas, growing along the ground as well as into the forest canopy. The dense growth and abundant leaves, which spring from the stems like small umbrellas, form a thick canopy just above the ground, and prevent sunlight from reaching other plants.




Photo: Randy Cyr, GREENTREE Technologies, www.invasive.org

Similarly, vines climbing up tree trunks spread out and surround branches and twigs, preventing most of the sunlight from reaching the leaves of the host tree. Loss of host tree vigor, evident within a few years, is followed by death a few years later. The added weight of vines makes infested trees susceptible to blow-over during storms. English ivy also serves as a reservoir for bacterial leaf scorch (Xylella fastidiosa), a plant pathogen that is harmful to native trees such as elms, oaks, and maples. English ivy is a popular plant, recommended by Cooperative Extension offices for use as a low maintenance alternative to lawns. It is widely used by homeowners, horticulturists, landscape contractors, parks departments and others desiring a fast-growing, low maintenance, evergreen groundcover. Once established at a site, English ivy can be expected to move beyond its intended borders into neighboring yards, parks and other lands, either by vegetative means or by seed.

**DISTRIBUTION IN THE UNITED STATES**: English ivy occurs in at least 26 states and the District of Columbia, where it is one of the most abundant and widespread invasive plants. Click here to see a distribution map.

**HABITAT IN THE UNITED STATES**: English ivy infests woodlands, forest edges, fields, hedgerows, coastal areas, salt marsh edges, and other upland areas, especially where some soil moisture is present. It does not grow well in extremely wet conditions and is often associated with some form of land disturbance, either human-caused or natural.

**BACKGROUND**: English ivy was probably first introduced to the US by European immigrants and is widely sold as an ornamental plant for landscapes throughout the US.

**BIOLOGY & SPREAD**: English ivy reproduces vegetatively and by seed, which is dispersed to new areas primarily by birds, including English house sparrows, European starlings, robins, Stellar jays, and cedar waxwings. New plants grow easily from cuttings or from stems making contact with the soil.

**For more information on English ivy, please contact:**
Sandra Diedrich, sddivy@teleport.com and http://www.noivyleague.com
Kris Johnson, Great Smoky Mountains National Park, Gatlinburg, TN
Sue Salmons, Rock Creek Park, Washington, D.C., sue_salmons@nps.gov
Jil Swearingen, National Park Service, Washington, D.C., jil_swearingen@nps.gov
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Southeast Exotic Pest Plant Council - Invasive Plant Manual, http://www.se-eppc.org/manual/
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service - Nonnative Invasive Plant of Southern Forests - http://www.invasive.org/eastern/srs/EI.html.

**SUGGESTED ALTERNATIVE PLANTS**: Attractive native vines are available that provide nectar for hummingbirds, butterflies, and other insects, serve as host plants for native insects, and provide food for many wildlife species. Vines native to the eastern U.S. include Allegheny pachysandra (Pachysandra procumbens), American or common bittersweet (Celastrus scandens), trumpet creeper (Campsis radicans), Virginia creeper (Parthenocissus quinquefolia), passionflower vine (Passiflora lutea), Dutchman's pipe (Aristolochia macrophylla), and native wisteria*

(Wisteria frutescens).

* If you wish to plant wisteria, make certain that it is the native species. Two commonly planted ornamental wisterias, Chinese wisteria (Wisteria sinensis) and Japanese wisteria (Wisteria floribunda), are exotic and aggressive invaders. Please consult the native plant society in your state for more information on species native to your particular area.

**AUTHORS**: Jil M. Swearingen, U.S. National Park Service, Washington, DC. Sandra Diedrich.

**REFERENCES:**

Collins, Christi. 1994. Some notes on Hedera helix The English ivy. The Nature Conservancy.
Dirr, Michael A. 1990. Manual of wody landscape plants: their identification, ornamental characteristics, culture, propagation and uses. Stipes Publ. Company, Champaign, IL.
Fernald, M.L. 1970. Gray's Manual of Botany. Eighth ed. D. Van Nostrand Co., New York, N.Y. p.1078.
Harty, Francis M. 1993. How Illinois kicked the exotic species habit. Pp. 195-209. In B.N. McKnight (ed.), Biological Pollution, Indiana Acad. Sci., Indianapolis, IN. 261 pp.
Lewis, W.H., and M. Elvin-Lewis. 1977. Meidical Botany: Plants Affecting Man's Health. John Wiley and Sons, Inc.
McElrone, A., Sherald, J. L. and Pooler, M. R. Identification of alternative hosts of Xylella fastidiosa in the Washington, DC area using nested polymerase chain reaction (PCR). J. Arboric. (In press)
Radford, A.E., H.E. Ahles, and C. Ritchie Bell. 1968. Manual of the Flora of the Carolinas. The University of North Carolina Press, Chapel Hill, NC. Pp. 758-9.
Randall, J. M., and Marinelli, J. 1996. Invasive Plants: Weeds of the Global Garden. Brooklyn Botanic Garden, Handbook #149. p. 7-11.
Salmons, S. 2000. Rock Creek Park Invasive Non-native Plant Mitigation Program, Final Report. January, 23 pp.
Sherald, J. L. and Kostka, S. J. 1992. Bacterial leaf scorch of landscape trees caused by Xylella fastidiosa. J. Arboric. 18: 57-63.
The Nature Conservancy. . English ivy: Element Stewardship Abstract. In: Wildland Invasive Species Program, Weeds on the Web. http://www.tnc.org
Thomas, L.K., Jr. 1980. The impact of Three Exotic Plant Species on a Potomac Island. National Park Service Scientific Monograph Series, no. 13. 179 pp.
Uphof, J.C.T. 1968. Dictionary of Economic Plants. J. Cramer Publishing Co., Lehre, Germany.
Virginia Native Plant Society. 1995. Invasive Alien Plant Species of Virginia: English ivy (Hedera helix).

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# English Ivy
# *Hedera helix* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance - Alien Plant Working Group -Weeds Gone Wild - Factsheet - English Ivy - http://www.nps.gov/plants/alien/fact/hehe1.htm - Author: Jil M. Swearingen and Sandra Diedrich - December 4, 2000**

Several effective methods of control are available for English ivy, including chemical and non-chemical, depending on the extent of the infestation, the amount of native vegetation on-site, and available time and labor.

## MANUAL AND MECHANICAL CONTROL:

**The Nature Conservancy - Weed Notes - English Ivy - http://tncweeds.ucdavis.edu/esadocs/hedeheli.html - Author: TunyaLee Morisawa - June 30, 1999**

Cutting is successful with persistence but does not always kill the plant. However, the use of cutting and then applying a herbicide may provide better control (see Chemical control section).

Using a shovel to remove plants provided immediate control with little regrowth. Weeding plants by hand or with pliers successfully allowed regeneration of most native species in Australia. Do not leave the pulled plants on the ground; they can continue to grow. If removal of the plants is not possible, place the pulled plants on a wooden platform to dry and decompose.

Immediately control English ivy that is growing up trees by cutting the vine at waist height, loosening the vine around the limbs and removing the roots. If the root can not be removed by hand, strip the bark and notch the exposed section of the vine. Paint on an undiluted herbicide such as glyphosate. If English ivy is growing on tree-ferns, take care that all pieces of the ivy are removed. The growth of H. helix can be sustained by the fibrous nature of the trunk.

**Plant Conservation Alliance - Alien Plant Working Group -Weeds Gone Wild - Factsheet - English Ivy - http://www.nps.gov/plants/alien/fact/hehe1.htm - Author: Jil M. Swearingen and Sandra Diedrich - December 4, 2000**

Vines growing as groundcover can be pulled up by hand, with some difficulty, and left on-site or bagged and disposed of as trash. Vines climbing up into the tree canopy are more difficult to manage. First, vines should be cut at a comfortable height to kill upper portions and relieve the tree canopy. A large screw driver or forked garden tool can be used to pry and snap the vines away from the tree trunks. Vines can be cut using an axe or with more difficulty, using a pruning saw. Rooted portions of vines will remain alive and should be pulled, and repeatedly cut. Because cutting will likely promote further growth from the base, vigilance is required to ensure long term control.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - English ivy - http://www.se-eppc.org/manual/HEHE.html - 2003**

Cutting: Cut climbing or trailing vines as close to the root collar as possible. This technique is feasible on small populations, as a pretreatment on

large impenetrable sites, in areas where herbicide cannot be used, or if labor resources are not sufficient to adequately implement herbicidal control. Severed vines may continue to live on tree bark or other porous surfaces for several growing seasons. English ivy will resprout unless cut so frequently that its root stores are exhausted.

Grubbing: This method is appropriate for small initial populations or environmentally sensitive areas where herbicides cannot be used. Using a pulaski or similar digging tool, remove the entire plant, including all roots and runners. Juvenile plants can be hand pulled depending on soil conditions and root development. Any portions of the root system not removed will potentially resprout.

Mulching: Mulching is an effective control on small infestations or in areas where herbicides cannot be used. Cover the entire infestation with several inches of mulch. This may include wood chips, grass clippings, hay or similar degradable plant material. Shredded or chipped wood may be the best option since hay and grass may potentially carry weed seeds. Covering the area with cardboard may improve the effectiveness and longevity of this method. The mulch should stay in place for at least two growing season and may need to be augmented several times.

## BIOCONTROL:

**Plant Conservation Alliance - Alien Plant Working Group -Weeds Gone Wild - Factsheet - English Ivy - http://www.nps.gov/plants/alien/fact/hehe1.htm - Author: Jil M. Swearingen and Sandra Diedrich - December 4, 2000**

There are no biological controls currently available for English ivy.

## CHEMICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group -Weeds Gone Wild - Factsheet - English Ivy - http://www.nps.gov/plants/alien/fact/hehe1.htm - Author: Jil M. Swearingen and Sandra Diedrich - December 4, 2000**

The systemic herbicide triclopyr (e.g., Garlon) is absorbed into plant tissues and carried to the roots, effectively killing the entire plant in place.

Foliar applications: From summer to fall, apply a 2.5% mixture of triclopyr amine (Garlon 3A) in water to the leaves or cut first, allow to regrow, and apply the same mix to new foliage. Herbicide will also be absorbed through the stem bark for additional effect.

Basal bark applications: A higher rate (15-30%) of triclopyr ester (Garlon 4) may also be applied to stems of vines growing up trees but there is a possibility that the herbicide will be absorbed into the host tree, depending on the thickness of the host tree's bark and the penetration of English ivy rootlets.

Because English ivy is an evergreen vine, and remains active during the winter, herbicide applications can be made to it any time of year as long as temperatures are above 55 or 60 degrees Fahrenheit for a few days. Fall and winter applications will avoid or minimize impacts to many native plant species. Repeat herbicidal treatments are likely to be needed and followup monitoring should be conducted to evaluate the success of treatments. Herbicidal contact with desirable plants should always be avoided. In areas where spring wildflowers or other native plants are interspersed, application of herbicides should be conducted prior to their emergence, or delayed until they have died back.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - English ivy - http://www.se-eppc.org/manual/HEHE.html - 2003**

Stump Treatment: Use this method in areas where vines are established within or around non-target plants, or where vines have grown into the canopy.

> Glyphosate: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of glyphosate and water to the cross-section of the stem. This procedure is effective at low temperatures (as low as 40°F) and may require a subsequent foliar application of glyphosate.
> Triclopyr: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of triclopyr and water to the cross-section of the stem. This procedure remains effective at low temperatures (<60°F) as long as the ground is not frozen. A subsequent foliar application may be necessary to control new seedlings.

Foliar Spray Method: Use this method to control large populations. It may be necessary to precede foliar applications with stump treatments to reduce the risk of damaging non-target species. Since English ivy is evergreen, the ideal time to spray is after surrounding vegetation has become dormant to avoid affecting non-target species.

> Glyphosate: Apply a 4% solution of glyphosate and water plus 0.5%-1% non-ionic surfactant to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. Glyphosate is a non-selective systemic herbicide that may kill non-target partially sprayed plants. Ambient air temperature should be above 65°F
> Triclopyr: If native grasses are intermingled with the ivy, triclopyr is preferred since it is selective to broadleaved plants. Apply a 2% solution of triclopyr and water to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. A 0.5% non-ionic surfactant is recommended in order to penetrate the leaf cuticle, and ambient air temperature should be above 65°F.

**The Nature Conservancy - Weed Notes - English Ivy - http://tncweeds.ucdavis.edu/esadocs/hedeheli.html - Author: TunyaLee Morisawa - June 30, 1999**

A wax layer on the leaves often prevents herbicides, especially hydrophilic compounds such as glyphosate, from permeating the leaves.

In container pots, two applications, one month apart, of 2,4-D (Weedar 64) applied at 1.1 kg/ha (1.0 lb/A) provided control of English ivy. Two

applications of glyphosate (Roundup) applied at 4.5 kg/ha (4.0 lb/A) effectively inhibited regrowth and provided some control. Regrowth but reduced shoot weight was observed with one treatment of 2,4-D and glyphosate at the rates stated above. The same observation was noted for one or two applications of glyphosate applied at a lower rate of 2.2 kg/ha (2.0 lb/A). Regrowth occurred with plants sprayed with one or two applications of Dicamba (Banvel) or triclopyr (Garlon) at the rate of 0.6 kg/ha (0.5 lb/A).

In another study, an application of glyphosate (25% solution) provided good control. Cutting (using a nylon cord weedeater to cut to the stem surface just before treatment) followed by a 25% solution of glyphosate also provided control of English ivy. Excellent control of H. helix that had been cut and then sprayed was achieved with a 2% solution of 2,4-D. A lower rate of glyphosate (2% solution) and cutting provided only slight control. Glyphosate only (2% solution) did not control English ivy. The herbicide triclopyr or mowing provided no control. Control evaluations were made 1 year post-treatment.

**Nonnative Invasive Plants of Southern Forests: a field guide for identification and Control: Bush honeysuckles - Gen. Tech. Rep. SRS-62. Asheville, NC, U.S. Department of Agriculture, Forest Service Research Station - http://www.invasive.org/eastern/srs/EI.html - Author: James H. Miller - 2003**

Recommended control procedures:

- Thoroughly wet all leaves (until runoff) with one of the following herbicides in water with a surfactant (July to October for successive years): Garlon 3A or Garlon 4 as a 3- to 5-percent solution (12 to 20 ounces per 3-gallon mix) or a glyphosate herbicide as a 2-percent solution (8 ounces per 3-gallon mix). Use a string trimmer to reduce growth layers and injure leaves for improved herbicide uptake. Cut large vines and apply these herbicides to cut surfaces.
- Or, apply Garlon 4 as a 20-percent solution in commercially available basal oil, diesel fuel, or kerosene (2.5 quarts per 3-gallon mix) with a penetrant (check with herbicide distributor) to large vines being careful to avoid the bark of the host tree.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Invasive Species Initiative - English Ivy - Comments on documents provided: December 16, 2004 - by: Joe Neal on article written by Jonathon Soll - http://tncweeds.ucdavis.edu/esadocs/hedeheli.html**

1)Joe Neal noted: "I would also suggest removing the recommendation to tank mix Scythe with Roundup. This combo seems to work fine on annual weeds, but its efficacy on perennial weeds is questionable. Limited research has been conducted with this combination applied to perennial weeds -- the one study I am aware of concluded that the combination was antagonistic and actually reduced the weed control.

2)Joe also noted: "My PhD dissertation was on the effects of glyphosate on woody ornamentals, and that research included English ivy. I conducted controlled studies in containers as well as field tests for English ivy control in the early 80s."

"In these trials the main conclusion was the critical importance of application timing. Treatments in the spring when plants had 2 to 4 newly expanded leaves provided the greatest control. Delaying treatment by only 6 weeks resulted in dramatically reduced control. Plants became increasingly more tolerant of glyphosate as the season progressed. See reference: Neal and Skroch 1985. Effects of timing and rate of glyphosate application on toxicity to selected woody ornamentals. J. Amer Soc Hort Sci 110:860-864."

"In the field applications I found that treatment with 2% by volume Roundup, in the spring when plants had 2 to 4 leaves, followed by another application about 6 weeks later (to re-growth) maximized control. When treated two years in a row in this fashion, we achieved over 95% control of established English ivy. " View the other resources related to this pest

Return to List

# DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers

## Orange daylily
## *Hemerocalis fulva* (L.) L.

**SOURCE:** The following information on this species is taken from Plant Invaders of Mid-Atlantic Natural Areas, http://www.nps.gov/plants/alien/pubs/midatlantic/ and invasive.org, www.invasive.org. Check the website links for future updates. The last update for the Plant Invaders information is 2002.

**OVERVIEW:** Common (or orange) daylily was introduced to the United States from Europe in the late 19th century. It is a very popular ornamental prized for its hardiness and variety -- there are now over 40,000 registered cultivars! Daylilies that have escaped from landscape plantings infest natural areas where they pose the greatest threat to meadows, floodplains, moist woods and forest edges. Daylilies reproduce by seed and also from thick, tuberous roots that grow rapidly to form dense clumps. Gardeners inadvertently spread daylilies by throwing away whole plants. They are difficult to control because of their thick tuberous roots.

**DESCRIPTION:** Leaves are long, grass-like, and bright green in color. Flowers are large, showy, and orange in color. Flowers may have spots or stripes. Many cultivars of daylily now exist and come in a wide variety of sizes and




Photo: Britt Slattery, U.S. Fish and Wildlife Service, www.forestryimages.org.



Photo: Dan Tenaglia, www.missouriplants.com, www.forestryimages.org.

**NATIVE ALTERNATIVES**: ox-eye sunflower (*Heliopsis helianthoides*), Canada lily (*Lilium canadense*), wood lily (*Lilium philadelphicum*), Turk's cap lily (*Lilium superbum*), three-lobed coneflower (*Rudbeckia triloba*)

**For more information on day lily, please contact:**
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Plant Invaders of Mid-Atlantic Natural Areas, http://www.nps.gov/plants/alien/pubs/midatlantic/
The Bugwood Network, MA-EPPC Invasive Plant List, http://www.invasive.org/maweeds.cfm
USDA - NRCS PLANTS Database, http://plants.usda.gov/

**Management and Control Information**

Return to List




## Species Management and Control Information

# Orange daylily
# *Hemerocallis fulva* (L.) L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## MANUAL AND MECHANICAL CONTROL:

**National Park Service and U.S. Fish & Wildlife Service - Plant Invaders of Mid-Atlantic Natural Areas - Common daylily - http://www.nps.gov/plants/alien/pubs/midatlantic/ - 2002**

Use spade/shovel to loosen soil and dig up root system. Re-sprouting may occur if entire root system is not removed.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Giant hogweed
## *Heracleum mantegazzianum* Sommier & Levier

**SOURCE:** The following information on this species is taken from the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Invasive Plant Fact Sheet for Giant hogweed, http://www.aphis.usda.gov/lpa/pubs/fsheet_faq_notice/fs_phhgweed.html. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is August 2002.

The U.S. Department of Agriculture's (USDA) Animal and Plant Health Inspection Service (APHIS) regulates giant hogweed (*Heracleum mantegazzianum*) as a noxious weed. This weed, which can be either biennial or perennial, grows to a height between 8 and 15 feet. Giant hogweed often has a taproot but can also have fibrous roots. It has hollow stems between 2 and 4 inches in diameter with dark reddish-purple splotches and pustulate coarse white hairs or bristles. The deeply incised compound leaves grow up to 5 feet in width. Hairs on the underside of the leaf are stiff, dense, and stubby. The large umbrella-shaped flower heads are up to 2 1/2 feet in diameter across a flat top with numerous small flowers. The small flower appears from mid-May through July. Giant hogweed produces flattened, oval-shaped fruit with a broad, rounded base and wide marginal ridges. This weed sprouts in the early spring from the roots or seed.




Photos (clockwise from left): Terry English, USDA - APHIS PPQ, www.forestryimages.org; Donna R. Ellis, University of Connecticut, www.forestryimages.org; Terry English, USDA-APHIS PPQ, www.forestryimages.org; USDA APHIS PPQ Archives, USDA APHIS PPQ, www.forestryimages.org.

Giant hogweed, which is native to the Caucasus, has been introduced into Europe, Great Britain, Canada, and the United States. Although it is listed as a Federal noxious weed and is illegal to bring into the United States or move across State lines, giant hogweed is known to occur in New York, Pennsylvania, and Washington. Reports of infestations have been received from Maine, Michigan, and Washington, DC. This weed has been planted as an ornamental in the United States and may have also been brought into this country for its fruit, used as a spice (golmar) in Iranian cooking.

Giant hogweed is likely to naturalize in many of the places where it has been first introduced. The plants thrive in many habitats but do particularly well where the soil has been disturbed, such as on wastelands, riverbanks, and along railroads. The weed prefers moist soil and partial shade, and can quickly dominate an area where the conditions are just right.

Both people and animals may have a sensitivity toward the giant hogweed's sap. Furocoumarins in the sap can cause a skin reaction known as photodermatitis. When photodermatitis occurs in people, their skin becomes sensitive to ultraviolet light in sunlight. This sensitivity can lead to long-lasting swelling and blisters. Contact with the eyes can cause temporary and sometimes permanent blindness.




Photo: USDA APHIS PPQ Archives, USDA APHIS, www.forestryimages.org.

APHIS' Federal noxious weed program is designed to prevent the introduction into the United States of nonindigenous invasive plants and to prevent the spread of newly introduced invasive plants within the United States. Noxious weed activities include exclusion, permitting, eradication of incipient infestations, survey, data management, public education, and (in cooperation with other agencies) integrated management of introduced weeds, including biological control.

**For more information on giant hogweed, contact:**
Dr. Alan Tasker, the Federal Noxious Weeds program coordinator, (301) 734-5708, Alan.V.Tasker@aphis.usda.gov
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Pennsylvania Department of Agriculture, Animal and Plant Health, Giant Hogweed, http://www.agriculture.state.pa.us/agriculture/cwp/view.asp?a=3&q=128383.
Plant Invaders of Mid-Atlantic Natural Areas, http://www.nps.gov/plants/alien/pubs/midatlantic/.
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA Animal and Plant Health Inspection Service (APHIS), http://www.aphis.usda.gov/lpa/pubs/fsheet_faq_notice/fs_phhgweed.html
USDA APHIS PPQ, AERO, Domestic Programs Pest Evaluation, Raleigh, NC, http://www.invasive.org/eastern/other/hogweed.html.
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA-NRCS PLANTS Database, http://plants.usda.gov/

**Management and Control Information**

Return to List




## Species Management and Control Information

# Giant hogweed
# *Heracleum mantegazzianum* Sommier & Levier

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

In Pennsylvania, Giant hogweed is a Pennsylvania Noxious Weed. If you find hogweed anywhere in the commonwealth, call the Toll Free Giant Hogweed Hotline at 1 (877) 464-9333. The PA Department of Agriculture can assist you in hogweed control and eradication.

## MANUAL AND MECHANICAL CONTROL:

**USDA Animal Plant Health Inspection Service - Pest Plant Quarantine - Domestic Programs Pest Evaluation - AERO - Raleigh, NC - http://www.invasive.org/eastern/other/hogweed.html - November 16, 2001.**

Mowing does not appear to be effective. The Pennsylvania Department of Agriculture has used a rototiller and planted a lawn seed mix to return areas to a natural state.

## BIOCONTROL:

**USDA Animal Plant Health Inspection Service - Pest Plant Quarantine - Domestic Programs Pest Evaluation - AERO - Raleigh, NC - http://www.invasive.org/eastern/other/hogweed.html - November 16, 2001.**

In Switzerland 12 phytophagous insect species found on native hogweed have been identified as possible candidates as biological control. Cattle and pigs are cited as possible biocontrol agents.

## CHEMICAL CONTROL*:

**USDA Animal Plant Health Inspection Service - Pest Plant Quarantine - Domestic Programs Pest Evaluation - AERO - Raleigh, NC - http://www.invasive.org/eastern/other/hogweed.html - November 16, 2001.**

The Pennsylvania Department of Agriculture recommends utilizing both pre and post emergent herbicides, plus competitive vegetation. PDA applies Pre-M at 1-1.5 oz./1000 s.f. in late-March. When germination of hogweed seed was noticed, Transline was incorporated at .5 oz./1000 s.f. along with Pre-M at the 1.5 oz/1000 s.f. rate. This continued until other vegetation started to grow. Later in the season, they used Drive at 1 oz./A. Post-emergent applications of 91% Thinvert, 7% Garlon 3A and 2% Transline are initiated in mid-April. These herbicide applications continue until snowfall covers the hogweed.

Previously, glyphosate has been considered the most effective herbicide, but it should be used with caution around desirable plants. The herbicides 2,4-D, TBA, MCPA, and dicamba are not effective on GHW roots. Rodeo (glyphosate) has been recommended in wet areas. Herbicides should be applied to large plants with protective clothing.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




## Dame's rocket
## Hesperis matronalis L.

**SOURCE:** The following information on this species is taken, in part, from the Virginia Tech Weed Identification Guide, Virginia Cooperative Extension, http://www.ppws.vt.edu/weedindex.htm and Randall, J.M. and J. Marinelli, editors, Invasive Plants: Weed of the Global Garden, Brooklyn Botanic Garden. Handbook #149. 1999. Specific species management and control information is provided elsewhere. Check the website links provided to check for future updates. The last update for the PCA fact sheet is December 3, 2001.

**NATIVE RANGE**: Europe

**DESCRIPTION**:Erect biennial or perennial that may reach 4 to 5 feet in height. Dame's Rocket is commonly viewed as a desirable wildflower-type plant that blooms in May or June in Virginia.







All Photos: Virginia Tech Weed ID Guide, http://www.ppws.vt.edu/scott/weed_id/

Lower leaves range from 3 to 8 inches in length and taper into a short petiole. Leaves become progressively smaller up the stem and are hairy on both sides. Upper leaves are also hairy but are without petioles or only have short petioles. All leaves are lanceolate in outline. Stems are erect, hairy, reaching 4 or 5 feet in height.




Flowers occur in showy clusters. Individual flowers are purple or white in color, 8 to 12 mm wide. Flowers are fragrant, especially in the evening. Fruit is a silique that is 2 to 4 inches long, 2 1/2 mm wide.

Some ironweeds like New York Ironweed (Vernonia noveboracensis) may resemble dame's rocket prior to flowering, however dame's rocket flowers very early in the season and has usually senesced by the time the ironweeds bloom later in the fall. Additionally, the leaves of dame's rocket are generally wider than those of New York ironweed.

**ECOLOGICAL THREAT**: Although it is not a large-scale invasive, dame's rocket can dominate moist areas of meadow, forest edge and alluvial woods to the exclusion of native plants. It spreads rapidly from seed.

**DISTRIBUTION IN THE UNITED STATES**: Dames rocket ranges across the United States and is found in all but about 9 of our extreme southern states.

**BACKGROUND**: Dame's rocket was introduced as a garden plant during the Colonial period, probably on the East Coast. It easily escaped cultivation and spread across most of the continent. It is still being used as a landscape plant and has become so common that many, mistakenly, believe that it is a native wildflower.

**BIOLOGY & SPREAD**: After spending the first half of its two-year life cycle as a rosette of leaves, dame's rocket plants develop rapidly the following spring into mature plants that flower, produce seed and die by mid summer. In the mid-Atlantic Coastal Plain region, seeds are produced in erect, slender, four-sided pods, called siliques, beginning in June. Siliques become tan and papery as they mature and contain shiny black seeds in a row. By late summer, the dame's rocket plants can be recognized only by the dead and dying stalks of dry, pale brown seedpods that may remain and hold viable seed throughout the late summer and fall.

**AUTHOR**:
Steve Young, New York State Natural Heritage Program.

**For additional information on dame's rocket**
: Invasive Plant Atlas of New England, http://webapps.lib.uconn.edu/ipane/browsing.cfm?descriptionid=114

**Management and Control Information**

Return to List




## Species Management and Control Information

# Dame's Rocket
# *Hesperis matronalis* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resources:

**Wisconsin Department of Natural Resources - Non-Native Plants - Dame's Rocket - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/dames.htm**

## GENERAL MANAGEMENT CONSIDERATIONS:

Dame's rocket has not been studied extensively. In fact, it is not yet widely recognized as an invasive plant in the Midwest. Consequently, this plant may not be recognized as a troublesome species until it is well-established as a formidable problem. Locating and removing plants immediately before seed sets is the best way to prevent the spread of dame's rocket. Be sure to check the contents of "wildflower" seed mixes for this species, and do not plant those that carry it.

## MANUAL AND MECHANICAL CONTROL:

Plants in urban gardens may not pose a problem, but any plant whose seed may escape to roadsides or woodlots should be eradicated or prevented from going to seed by cutting the flower heads after they bloom. Pulling may need to be done for several years to remove new plants established from the seed bank. Pulling or use of a dandelion digger is most effective when the soil is moist is effective. If plants are pulled when in bloom, they should not be placed in compost piles, as the seeds may still ripen and spread. Flower-heads should be bagged for landfill, or dried and burned where permissible. Where there is sufficient leaf litter or other fuel, burning has been found to be an effective control method.

## CHEMICAL CONTROL:

Selectively applying a broadleaf herbicide like glyphosate to seedlings according to label recommendations may also be an effective means of control. To avoid damaging adjacent native vegetation, apply herbicide in late fall when the rosettes are still green.

## OTHER CONSIDERATIONS:

NOTE: See garlic mustard (Alliaria petiolata) Management and Control for other possible control options.

Return to List




# Purple Loosestrife
# Lythrum salicaria L.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is March, 2005.

**NATIVE RANGE**: Eurasia; throughout Great Britain, and across central and southern Europe to central Russia, Japan, Manchuria China, southeast Asia and northern India

**DESCRIPTION**: Purple loosestrife is an erect perennial herb in the loosestrife family, with a square, woody stem and opposite or whorled leaves. Leaves are lance-shaped, stalkless, and heart-shaped or rounded at the base. Plants are usually covered by a downy pubescence.



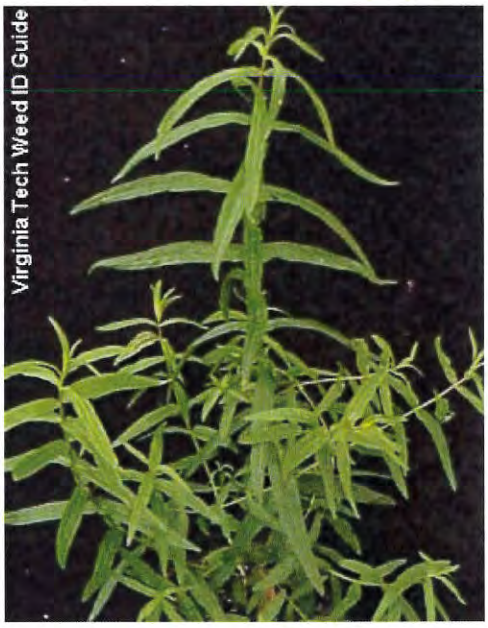




Photo: Linda Wilson, University of Idaho, www.forestryimages.org

Loosestrife plants grow from four to ten feet high, depending upon conditions, and produce a showy display of magenta-colored flower spikes throughout much of the summer. Flowers have five to seven petals. Mature plants can have from 30 to 50 stems arising from a single rootstock.




Photo: Linda Wilson, Univ. of Idaho, www.forestryimages.org.



Photo: Illinois Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html

**ECOLOGICAL THREAT**: Purple loosestrife adapts readily to natural and disturbed wetlands. As it establishes and expands, it outcompetes and replaces native grasses, sedges, and other flowering plants that provide a higher quality source of nutrition for wildlife. The highly invasive nature of purple loosestrife allows it to form dense, homogeneous stands that restrict native wetland plant species, including some federally endangered orchids, and reduce habitat for waterfowl.




Photo: Agriculture and Agri-Food Canada Archives, Agriculture and Agri-Food Canada, www.forestryimages.org.

**DISTRIBUTION IN THE UNITED STATES**: According to the U.S. Fish and Wildlife Service, purple loosestrife now occurs in every state except Florida.

**HABITAT IN THE UNITED STATES**: Purple loosestrife is capable of invading many wetland types, including freshwater wet meadows, tidal and non-tidal marshes, river and stream banks, pond edges, reservoirs, and ditches.

**BACKGROUND**: Purple loosestrife was introduced to the northeastern U.S. and Canada in the 1800s, for ornamental and medicinal uses. It is still widely sold as an ornamental, except in states such as Minnesota, Wisconsin, and Illinois where regulations now prohibit its sale, purchase and distribution.

**BIOLOGY & SPREAD**: Purple loosestrife enjoys an extended flowering season, generally from June to September, which allows it to produce vast quantities of seed. The flowers require pollination by insects, for which it supplies an abundant source of nectar. A mature plant may have as many as thirty flowering stems capable of producing an estimated two to three million, minute seeds per year.

Purple loosestrife also readily reproduces vegetatively through underground stems at a rate of about one foot per year. Many new stems may emerge vegetatively from a single rootstock of the previous year. "Guaranteed sterile" cultivars of purple loosestrife are actually highly fertile and able to cross freely with purple loosestrife and with other native Lythrum species. Therefore, outside of its native range, purple loosestrife of any form should be avoided.

**SUGGESTED ALTERNATIVE PLANTS**: Native species of Liatris (blazing star) have showy pink-purple flower spikes and are an important nectar source for many native species of butterflies and other insects.

**AUTHOR:**
Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on garlic mustard, please contact:**
Cornell University Non-indigenous Plant Species Program http://www.invasiveplants.net
Delaware River Invasive Plant Partnership, http://www.paflora.org/DRIPP.html
Illinois Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Ohio Department of Natural Resources, Fact Sheets, http://www.dnr.state.oh.us/dnap/invasive/4loosestrife.htm
Southeast Exotic Pest Plant Council, Invasive Plant Manual, http://www.se-eppc.org/manual/
The Bugwood Network, http://www.invasive.org/
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
Virginia Natural Heritage Program http://www.state.va.us/~dcr/dnh/invlyth.htm
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm

**REFERENCES:**
Heidorn, R. and B. Anderson. 1991. Vegetation management guideline: purple loosestrife (Lythrum salicaria L.). Natural Areas Journal 11:172-173.
LaFleur, A. 1996. Invasive plant information sheet: purple loosestrife. The Nature Conservancy, Connecticut Chapter.
Malecki, R.A. (et al.). 1993. Biological control of purple loosestrife (Lythrum salicaria). BioScience 43 (10):680-686.
The Nature Conservancy. Purple Loosestrife: Element Stewardship Abstract. In: Wildland Weeds Management & Research Program, Weeds on the Web.
Thompson, Daniel Q., Ronald L. Stuckey, Edith B. Thompson. 1987. Spread, Impact, and Control of Purple Loosestrife (Lythrum salicaria) in North American Wetlands. U.S. Fish and Wildlife Service. 55 pages.
Virginia Department of Conservation and Natural Resources. 1995. Invasive Alien Plant Species of Virginia: Purple Loosestrife (Lythrum salicaria).

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Purple loosestrife
# *Lythrum salicaria* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/ploosestrife.html - Vol.1, No. 17 - Authors Randy Heidorn - February 6, 1990.**

The chances of success are best with the smallest infestations, and methods of control are determined by the size of the infestation. Early diagnosis is critical. Decisions on control methods must be balanced with the chances of success and the poten-tial damage caused by the treatment method.

Potential loosestrife habitat should be searched annually during late July and August for the plant. Early detection is the best approach!

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Purple loosestrife - http://tncweeds.ucdavis.edu/esadocs/lythsali.html - Author: J. Bender; update by Jay Rendall - November 1, 1987.**

Once purple loosestrife becomes established in a wetland it displaces endemic vegetation through rapid growth and heavy seed production (Rawinski 1982). L. salicaria has a detrimental impact on native wetland vegetation and associated wildlife. Important wildlife food plants such as cattails and pondweed are displaced or shaded out as L. salicaria expands across a wetland. If purple loosestrife is left unchecked, the wetland eventually becomes a monoculture of loosestrife (Rawinski 1982). The invasion of L. salicaria leads to a loss of plant diversity, which also leads to a loss of wildlife diversity.

Management objectives may include eradicating populations, containing populations or preventing establishment. Monitoring should be used to track the accomplishment of these objectives.

The best time to search for purple loosestrife is in July and August when the plants are blooming. The bright magenta flowers are easy to spot at a great distance. Aerial surveys can be used to note the yearly position of large populations. An advancing or receding boundary would be identifiable from air photos. Ground surveys are more feasible for tracking small populations and finding newly established populations. Look for seedlings in June.

The following individuals are involved in public awareness campaigns or wetland surveys:

John Schwegman, Director, Botany Program, Illinois Dept. Conservation, Springfield, Illinois 62706.

Rich Henderson, Consultant, Natural Areas Management, 2845 Timberlane Verona, Wisconsin 53593.

Noel Cutright, Purple Loosestrife Task Force, 3352 Knollwood Rd., West Bend, Wisconsin 53095.

Purple loosestrife Survey-Nevin, Wisconsin Dept. Natural Resources, P.O. Box 7921, Madison, Wisconsin 53791.

Jay Rendall, Coordinator, Purple Loosestrife Program, Minnesota Dept. Natural Resources, Box 25, 500 Lafayette Rd., St. Paul, MN 55155.

Bonnie Harper, Purple Loosestrife Coalition, 14375 Valley View Rd., Apt. F, Eden Prairie, MN 55344.

Several control methods have been attempted with varying degrees of success. Natural area managers must determine their objectives first. Is it more feasible to contain or control populations of purple loosestrife? Large populations extending over three acres or more will be difficult if not impossible to completely eradicate using presently known methods. These large populations should be contained at their present position. Preventing the expansion can be accomplished through hand-pulling new plants along the periphery or spraying herbicide on plants extending beyond the main body of the population. Smaller populations can be controlled through eradication. Populations up to three acres can be cleared with herbicides or hand-pulled, depending upon the size of the work crew and time available.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Purple loosestrife- http://www.nps.gov/plants/alien/fact/lysa1.htm - Authors: Jil M. Swearingen - March 28, 2005.**

Small infestations of young purple loosestrife plants may be pulled by hand, preferably before seed set.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Fact Sheet - Purple loosestrife - http://www.dnr.state.oh.us/dnap/invasive/4loosestrife.htm -January 8, 2003.**

Small infestations of purple loosestrife can be removed by hand. The entire root system must be removed from the ground. All plant material should be bagged and removed from the area to eliminate re-sprouting. Larger populations are harder to control using mechanical means. Mowing should not be used because it can increase the spread of the population by dispersing seeds and exposing the seed bank.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Purple loosestrife - http://www.se-eppc.org/manual/loosestrife.html - 2003**

Hand Pulling: In areas that contain less than 100 plants, younger plants (1-2 years old) can be hand-pulled. Plants more than 2 years old should be dug out with special care to include the entire rootstock. Removal activities should take place before flowering to ensure that seeds are not dispersed during the disturbance. All plant parts should be carefully bagged, removed from the site, and placed in approved landfills or preferably burned. Any plant fragment that escapes proper disposal could spread purple loosestrife on your control site or along your travel route. In addition, all clothing, boots, and equipment should be properly cleaned to ensure that no seeds are transported. If feasible, native plants should be restored to the control area by seeding or planting. This reestablishment of vegetation will deter new loosestrife seedling development through competition. Do not cut or mow purple loosestrife. These methods will simply increase the spread of plants since they can sprout vegetatively.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Purple loosestrife - http://tncweeds.ucdavis.edu/esadocs/lythsali.html - Author: J. Bender; update by Jay Rendall - November 1, 1987.**

PULLING: Hand-removal is recommended for small populations and isolated stems. Ideally, the plants should be pulled out before they have set seed. The entire rootstock must be pulled out since regeneration from root fragments is possible. Be sure to minimize disturbances to the soil and native vegetative cover. Remove uprooted plants and broken stems from the area since the broken stems can resprout (Rawinski 1982).

REPLACEMENT: Replacement control has been attempted in several wildlife refuges (Balogh 1985, Rawinski 1982). Rawinski (1982) sowed Japanese millet (Echinochloa frumentacea) with L. salicaria and found that the millet seedlings outcompeted the loosestrife seedlings. The millet must be planted immediately after marsh drawdown has occurred. Balogh (1985) found that Japanese millet does not regenerate well and would have to be replanted every year. Balogh (1985) attempted a replacement treatment using native seed. Polygonum lapathifolium was seeded with purple loosestrife and the Polygonum outcompeted the loosestrife. However, the following spring L. salicaria would start growing first due to its overwintering rootstock. Replacement methods would have a very limited application within a natural area, but they may be useful to control or contain loosestrife populations on buffer property.

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/ploosestrife.html - Vol.1, No. 17 - Authors Randy Heidorn - February 6, 1990.**

Effort in areas with individual plants and cluster of up to 100 plants Younger plants (1-2 years old) can be hand-pulled. Do not pull after flowering because this will scatter seed. Older plants, especially those in bogs or in deep organic soils, can be dug out. Roots of older plants can be "teased" loose with a hand cultivator. Bag and remove the plants from the site. Failure to place the removed plants in a bag could result in spread-ing the plant along your exit route because fragments may be dropped. Dispose of the plant by burning (preferable) or in an approved landfill. Follow-up treatments are recommended for three years after the plants are removed. Clothing, equipment and personnel should be cleaned to insure no seeds are spread on them, if seeds were present on plants.

If the above control method is not feasible in areas with relatively small infestations, spot application of glyphosate herbicides can be used as described below.

Cutting purple loosestrife and subsequently flooding the area so that cut plant stalks are completely immersed has controlled purple loosestrife in at least one case. However, flooding may encourage the spread of purple loosestrife if seed are present in the soil. Artificial flooding should not be used in high-quality natural communities with an intact natural flooding regime.

## BIOCONTROL:

**Van Driesche, R., et al., 2002, Biological Control of Invasive Plants in the Eastern United States, USDA Forest Service Publication FHTET-2002-04, 413 p. http://www.invasive.org/eastern/biocontrol/11PurpleLoosestrife.html.**

Based on results indicating a potential wider host range, the gall midge B. salicariae was not proposed for introduction (Blossey and Schroeder, 1995). After review by the Technical Advisory Group, it was determined that further invasion by L. salicaria is considered a greater threat to the native L. alatum and D. verticillatus than potential attack by the leaf beetles or the root feeder, and releases were approved. Initial introductions in eastern North America occurred in Virginia, Maryland, Pennsylvania, New York, Minnesota, and southern Ontario in August, 1992 (Hight et al., 1995). Predictions that at high population densities beetles might nibble at other species (Blossey et al., 1994a, b; Blossey and Schroeder, 1995) were confirmed (Corrigan, 1998; Blossey et al., 2001b), but attack was transient and restricted to newly emerging beetles.

Approval to introduce the flower-feeding weevil N. marmoratus was granted followed by introductions in New York and Minnesota in 1994. Additional releases occurred in New Jersey in 1996. The seed-feeding weevil N. brevis, while approved for introduction, was not released into North America, due to the presence of a nematode infection. This infection appeared benign for N. brevis, however, due to the potential for non-target effects of the nematode after introduction into North America, only disease free specimens should be introduced, which, at present, effectively precludes the introduction of N. brevis.

All four introduced species have successfully established in North America. The two Galerucella species are established in Maine, Massachusetts, Connecticut, Rhode Island, Vermont, New Jersey, New York, New Hampshire, Maryland, Delaware, Virginia, West Virginia, Pennsylvania, Ohio, Indiana, Tennessee, Michigan, Illinois, Wisconsin, Minnesota, Kansas, and Iowa. The species have spread up to 5 km from the original release sites and G. calmariensis appears to be more successful than G. pusilla. The secretive nature of H. transversovittatus makes assessments of its status difficult. Releases have occurred throughout the United States but establishment (attacked roots) is confirmed only for Colorado, Maryland, Pennsylvania, New York, Indiana, Minnesota, New Jersey, Michigan, and Virginia. The flower-feeding weevil now occurs in New York, New Jersey, Colorado, and Minnesota, and populations are expanding.

At several release sites complete defoliation of large purple loosestrife stands (many hectares) has been reported with local reductions of more than 95% of the biomass (Fig. 8). Such outcomes are currently restricted to some of the earlier release sites but similar observations have been made in Rhode Island, Connecticut, New York, Indiana, Michigan, Illinois, Minnesota, and Canada.

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Purple loosestrife- http://www.nps.gov/plants/alien/fact/lysa1.htm - Authors: Jil M. Swearingen - March 28, 2005.**

While herbicides and hand removal may be useful for controlling individual plants or small populations, biological control is seen as the most likely candidate for effective long term control of large infestations of purple loosestrife. As of 1997, three insect species from Europe have been approved by the U.S. Department of Agriculture for use as biological control agents. These plant-eating insects include a root-mining weevil (Hylobius transversovittatus), and two leaf-feeding beetles (Galerucella calmariensis and Galerucella pusilla). Two flower-feeding beetles (Nanophyes) that feed on various parts of purple loosestrife plants are still under investigation. Galerucella and Hylobius have been released experimentally in natural areas in 16 northern states, from Oregon to New York. Although these beetles have been observed occasionally feeding on native plant species, their potential impact to non-target species is considered to be low.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Fact Sheet - Purple loosestrife - http://www.dnr.state.oh.us/dnap/invasive/4loosestrife.htm -January 8, 2003.**

Several species of insects are being studied for their effectiveness in the control of purple loosestrife. A species of weevil (Hylobius transversovittatus) lays eggs in the stem and upper root system of the plant and as the larvae develop, they feed on root tissue. Two species of leaf-eating beetles (Galerucella calmeriensis and G. pusilla) and a weevil (Nanophyes marmoratus) that feeds on flowers and stresses the plant are being released into areas of high purple loosestrife density and are being monitored. Since 1994, the Ohio Division of Wildlife has introduced these insects into 13 areas. Although this method will not eradicate the species, it may create a more tolerable population level that will stabilize over time.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Purple loosestrife - http://www.se-eppc.org/manual/loosestrife.html - 2003**

Long-term studies on the effectiveness of biological controls are being conducted at the New York Cooperative Wildlife Research Unit at Cornell University. Several phytophagous insects which specifically feed on purple loosestrife in Europe have under-gone a series of intensive lab and field testing. Three beetles-two leaf eaters, and one root miner-have been approved for release in the U.S. Experimental importation of these insects has been made in the northeast. Success of these efforts could pave the way for the use of biological controls to manage purple loosestrife in a permanent, cost-effective, and environmentally sound way. There is significant concern about other native species of the genus Lythrum that may also be fed upon, although to a lesser degree, by these insects. Since other control measures may harm a variety of non-target plant species, this non-target feeding may occur at a level which is preferable to alternative control techniques.

## CHEMICAL CONTROL:

**Plant Conservation Alliance, Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Purple loosestrife- http://www.nps.gov/plants/alien/fact/lysa1.htm - Authors: Jil M. Swearingen - March 28, 2005.**

For older plants, spot treating with a glyphosate type herbicide (e.g., Rodeo for wetlands, Roundup for uplands) is recommended. These herbicides may be most effective when applied late in the season when plant are preparing for dormancy. However, it may be best to do a mid-summer and a late season treatment, to reduce the amount of seed produced.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Fact Sheet - Purple loosestrife - http://www.dnr.state.oh.us/dnap/invasive/4loosestrife.htm -January 8, 2003.**

Herbicides can be used effectively to control small populations of purple loosestrife. Only herbicides permitted for wetland use, such as Accord® or Glypro®, may be used. By eliminating all the plants in an area, the soil is exposed for the immense purple loosestrife seed bank to germinate. Spot application of herbicide can help limit this problem. The most species specific way to apply herbicide is by cutting and treating the stems. Foliar spray can be used by applying herbicide after the period of peak bloom, in late August. Any control method should be followed up on a yearly basis to catch any missed plants or new sprouts. Certain broadleaf specific herbicides, such as Garlon 3A®, which do not harm monocot species (grasses and sedges) that typically occur in wetlands, can also be used.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Purple loosestrife - http://www.se-eppc.org/manual/loosestrife.html - 2003**

Cut Stump Treatment: In areas that contain more than 100 plants, a spot application of a glyphosate herbicide (one that is approved for use in and near water) is recommended. Individual purple loosestrife plants should be cut about 15 cm (6 inches) above the ground. A 20-30% solution of glyphosate and water should be applied directly to the cut surface either by a wick or injection into the stem.

Foliar Spray Method: If purple loosestrife covers a large area, a foliar spray can be applied using a 2% glyphosate solution and water plus 0.5% non-ionic surfactant. To be most effective herbicide should be applied just when plants have begun flowering. Where feasible, flower heads should be cut, bagged, and removed from the site before application to prevent the production of seed. Glyphosate is a non-selective herbicide, and extreme care must be taken to avoid contact with non-target plant species. The restoration of sites depends on these non-target species as they recolonize the area after the purple loosestrife is eliminated.

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Purple loosestrife - http://tncweeds.ucdavis.edu/esadocs/lythsali.html - Author: J. Bender; update by Jay Rendall - November 1, 1987.**

CHEMICAL: The herbicide glyphosate is most commonly used to control L. salicaria. Glyphosate is available under the trade names RoundupTM and RodeoTM, manufactured by Monsanto. Roundup cannot be used over water. Another formulation of glyphosate known as Rodeo contains a non-ionic surfactant and has been approved for use over water. Ortho X-77 is the non-ionic surfactant recommended for use with Rodeo, but several other non-ionic surfactants were cleared for use with Rodeo in 1985 (Balogh 1985).

The major disadvantage in using Rodeo is that glyphosate is a non-specific systemic. Broadcast spraying of non-selective herbicides kills all of the vegetation and may result in an increase in loosestrife density because of seed germination following the removal of competing perennial vegetation (Minnesota DNR 1987). Spot application of Rodeo directly onto L. salicaria would ensure that no large holes would appear in the marsh vegetation and that competition would be unaffected. The safest method of applying glyphosate herbicide is to cut off all stems at about 6 inches and then paint or drip onto the cut surface a 20-30% solution (Henderson 1987).

Spraying should be done after the period of peak bloom, usually late August (Balogh 1985, Rawinski 1982). One to two percent solutions of Rodeo have been recommended as sufficient to kill L. salicaria (Henderson 1987, Minnesota DNR 1987, Balogh 1985, Thrune pers. comm.). Work done by Jim Reinartz at the U.W.-Milwaukee Field Station indicates it is best to spray no more than 25-50% of a plant's foliage (Henderson 1987). This will help protect against overspraying which might damage adjacent vegetation.

It is critical that any control effort be followed up the same growing season and for several years afterwards since some plants will be missed, new seedlings may sprout from the extensive seed bank, and a few plants will survive the low-dosage treatment (Henderson 1987, Minnesota DNR 1987). Higher dosage and careless application, however, inevitably kills more surrounding vegetation and leads to establishment of loosestrife seedlings (Minnesota DNR 1987).

For larger infestations where spot application of glyphosate is not practical, broadleaf herbicides can be used. They have the advantage of not harming monocot species, which are the dominants in most wetland types. Broadleaf herbicides (2,4-D based) can be effective on loosestrife if applied in late May or early June (Henderson 1987). The disadvantage of treating early in the season is that purple loosestrife plants are easily overlooked when not in flower. A combination of 2,4-D and dicamba has been used on a limited basis in western irrigation ditches (Jackson pers. comm.). The EPA has approved a 1:1 tank mix of these two products. Once L. salicaria has reached 10-15% of its mature growth, it can be sprayed with good results. To ensure complete coverage and compensate for spotty application, repeat the treatment once during the growing season (Jackson pers. comm.).

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/ploosestrife.html - Vol.1, No. 17 - Authors Randy Heidorn - February 6, 1990.**

Effort in areas with clusters in excess of 100 plants (up to 4 acres in size) Spot application of a glyphosate herbicide to individual purple loosestrife plants is the recommended treatment where hand pulling is not feasible. Glyphosate is available under the trade names Roundup and Rodeo, products manufactured by Monsanto. Only Rodeo is registered for use over open water. Herbicides only may be applied according to label directions and by licensed herbicide applicators or operators when working on publc properties. Glyphosate is nonselective so care should be taken not to let it come in contact with nontarget species. Glyphosate application is most effective when plants have just begun flowering. Timing is crucial, because seed set can occur if plants are in mid-late flower. Where feasible, the flower heads should be cut, bagged, and removed from the site before application to prevent seed set. Roundup should be applied by hand sprayer as a 1-1/2% solu-tion (2 oz. Roundup/gallon of clean water). Rodeo should also be applied as a 1-1/2% solution (2 oz. Rodeo/gallon clean water) with the addition of a wetting agent, as specified on the Rodeo label.

Another option is to apply glyphosate twice during the growing season. Foliage should be sprayed as described above, once when flowering has just started and a second time 2-3 weeks later. With this procedure control is likely more effective, because plants are not allowed to set seed and those missed because they were not flowering the first time are treated the second time.

Excessive application of herbicide (causing dripping from the plant) can kill desirable plants under the loosestrife. These plants, left unharmed, will be important in recolnizing the site after the loosestrife has been controlled. If the desirable plants are killed, the vigorously resprouting and growing purple loosestrife seeds present in the soil will fill the void. Since purple loosestrife is usually taller than the surround-ing vegetation, application to the tops of plants alone can be very effective and limit exposure of nontarget species. Complete coverage is not required to affect

control.

The herbicide should be applied while backing away from treated areas to avoid walking through the wet herbicide. Equipment, clothing and personnel should be cleaned completely before entering other uninfested sensitive areas, if seeds were present in the treated area. It will be necessary to treat the same area again annually until missed plants and plants originating from the seed bank are eliminated.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Species Management Summary (ESA or Element Stewardship Abstract): Purple loosestrife - http://tncweeds.ucdavis.edu/esadocs/lythsali.html - Author: J. Bender; update by Jay Rendall - November 1, 1987.**

Management Research Programs:
A research project in Wisconsin includes investigations on different methods of control and different herbicide treatments. The ecology of L. salicaria including seed bank buildup is also under investigation. Contact: Rich Henderson, Consultant-Natural Area Management. 2845 Timberlane, Verona, Wisconsin 53593.

Hand cutting purple loosestrife and fertilizing cattails under varying degrees of wetness is being studied at Indiana Dunes National Lakeshore. Contact: Ron Heibert, Chief, Division of Science, 1100 N. Mineral Springs Rd., Porter, Indiana 46304.

Research on the effectiveness of various chemical controls will be conducted in Illinois, pending funding. Contact: John Schwegman, Director, Botany Program, Illinois Dept. Conservation, Springfield, Illinois 62706.

A research project funded by the Metropolitan Council (MN) is being conducted by Hennepin County Park Reserve. Chemical control techniques are to be evaluated for 2 years in control plots. Contact: Tom Hollenhorst, Hennepin County Park Reserve, 3800 Co. Rd. 24, Maple Plain, MN 55359.

The Minnesota Legislative Commission on Minnesota Resources (LMCR) has funded a comprehensive control program over a two year period. The program will inventory purple loosestrife in Minnesota, keep abreast of current contol methods and research, implement a prioritized control program, monitor environmental impact and effectiveness of control, promote public awareness campaigns, and coordinate agencies control efforts within the state. Contact: Jay Rendall, Coordinator, Purple Loosestrife Program, MN Dept of Natural Resources, Box 25, 500 Lafayette Rd, St. Paul, MN 55155.

Management Research Needs:
Biological control methods should be a priority for research. Repeated chemical treatments are costly and the long-term effects on natural systems are not fully understood. Preliminary investigations in Europe have revealed several host-specific insects that keep L. salicaria in check. Further research is warranted. Research is needed to assess the potential productivity of the seed bank. How extensive is the seed bank in a wetland in comparison to the size of the above-ground population? What is the rate of seed buildup? Can the age of a seed bank be determined? What is the viability of purple loosestrife seed? More research is needed on herbicide treatments that will give the most selective application with the least impact to the surrounding competitive vegetation, i.e. wick applications, etc. Available information suggests that research on mechanical treatments will not yield helpful results.

The following individuals are familiar with L. salicaria and can suggest control strategies:

Dottie Thompson, Horicon National Wildlife Refuge, Rt. 2, Mayville, Wisconsin 53050. (414) 387-2658.

Rich Henderson, Consultant, Natural Area Management, 2845 Timberlane, Verona, Wisconsin 53593. (608) 845-7065.

John Schwegman, Director, Botany Program, Illinois Dept. Conservation, Springfield, Illinois 62706.

Tom Jackson, Leader, Field Research Station, U.S. Fish and Wildlife Service, P.O. Box 25007, Denver, Colorado 80225.

Jay Rendall, Coordinator, Purple Loosestrife Program, MN Dept of Natural Resources, Box 25, 500 Lafayette Rd, St. Paul, MN 55155.

**Illinois Vegetation Management Guideline - http://www.inhs.uiuc.edu/chf/outreach/VMG/ploosestrife.html - Vol.1, No. 17 - Authors Randy Heidorn - February 6, 1990.**

Effort in areas with large monocultures (greater than 4 acres in size) For large purple loosestrife populations, an assessment should be made to determine if the loosestrife can be eradicated with available resources. If it can not be controlled, then efforts should be placed on keeping the loosestrife out of the highest quality areas. Applying glyphosate from a vehicle mounted sprayer is usually necessary in areas with extensive stands of purple loosestrife. The most effective control can be achieved by beginning treatment at the periphery of large patches and working towards the center in successive years. This allows peripheral native vegetation to reinvade the treated area as the loosestrife is eliminated.

FAILED OR INEFFECTIVE PRACTICES
Mowing, burning, and flooding have proven largely ineffective. A single known exception is cutting followed by flooding as described above. Mowing and flooding can actually contribute to further spread of the species by disseminating seed and cut plant stems. Do not mow because cut parts may re-root. No biological control methods that are feasible in natural areas are known.

**The Nature Conservancy - Invasive Species Initiative - Comments from Stewards - Purple loosestrife - http://tncweeds.ucdavis.edu/esadocs/lythsali.html - Compiled by Mandy Tu. 2000.**

Techniques from TNC Stewards for the eradication of Lythrum salicaria (purple loosestrife) and Phragmites australis (common reed/Phrag) in wetlands - Edited by Mandy Tu, Wildland Invasive Species Program, The Nature Conservancy, Dept. of Vegetable Crops & Weed Sciences, University of California at Davis

This past summer (2000), Betsy Lyman, the Assistant Director of Science & Stewardship for TNC's Pennsylvania Field Office, sent out a call in both Stew News (an in-house TNC publication) and on TNC's Invasive Species Listserve requesting control information for Lythrum salicaria and Phragmites australis. There was a large response to these questions, and we gathered this information, and lightly edited it. If you have either of these weeds on your preserve, you may benefit from reading about others' control experiences.

Contents:
I. Betsy's original inquiry and questions.
II. Personal accounts/Case studies
A. Excellent control of P. australis. L. salicaria eradication still in progress. (Indiana -- C. O'Leary)
B. Great success in controlling P. australis. Control of L. salicaria promising after 2 years. (Indiana -- M. Norris)
C. Excellent, but labor-intensive, control of P. australis. Potential successful control of L. salicaria. (New Jersey -- M. Morris)
D. Effective control of L. salicaria. (Washington -- M. Summers)
III. Methodologies
A. Application of herbicide to P. australis and L. salicaria. (Michigan -- J. McGowan-Stinski, D. Borneman, K. Condict)
B. Adaptive management for P. australis control. (New York -- B. Patterson) IV. Suggested references V. Contact information

Betsy's original inquiry and questions

Many Lythrum salicaria questions (Pennsylvania)

Pennsylvania has a fen complex which contains federally and state-listed plants and reptiles. We have both Phragmites australis and Lythrum salicaria (purple loosestrife) in several of these fens. We have used groups of volunteers to cut the offending plants (for Phragmites, we cut in mid-June and mid-September; for loosestrife, we cut in late July/early August when it is blooming). However, the US FWS does not like the idea of hordes of volunteers trampling through the fens, since they could easily harm the nests of the endangered turtle. So we are looking into using herbicides-i.e. Rodeo (glyphosate).

1) Are there any studies on the effects of glyphosate applications on turtles?
2) What methods (manual, herbicides, etc.) have you used that were effective in controlling the weeds, but which have not negatively impacted the native vegetation? What worked best?
3) Have you done your control work stepwise (i.e. a little bit each year) or have you done the herbiciding all at once?
4) Have you followed up with any planting of native plants?
IIA. Case study: Control efforts in Indiana

We have worked with those two wicked invasive plants in fens and other wetlands. We have had great success with killing Phragmites australis. We use 1.5% Rodeo applied from backpack sprayers with 5 foot wand extensions. (The long wands are necessary to reach the tops of the plant). Our best results have been after late-season spraying, close to, but before, the plants senesce. In Indiana this is the first 3 weeks of September. This technique has been used in large monotypic stands (5-20 acres) where the understory is degraded (mostly Phragmites duff) and we have measured over 97% mortality after the first year of spraying. We then burn the duff the next spring to make follow up easier and to allow the native seed bank to germinate.

In areas where the Phragmites is occasional and intermingled with the natives, we use our best discretion - either carefully spray small stands or hand-cut the stems and drip 25% Rodeo into the stump. Hand-cutting is very time consuming if the stands are really big and is less effective (50-75% mortality), but there is no overspray. We have found that workdays with a lot of hands makes it less tedious and more practical to achieve results. If you decide to spray, which is much more time efficient, the sprayer should be experienced, and overspray can be controlled pretty well.

I have been less successful with purple loosestrife (Lythrum salicaria). My opinion is that hand-cutting and pulling are a complete waste of time and lead to great frustration. Foliar spray with 1.5-3.0% Rodeo is our current technique. In two years of intensive work, we have pushed purple loosestrife from a dominant plant to an occasional plant, but eradication is still many years away. This is due to the incredible seed bank. These plants are tough and sometime require a second application of herbicide to kill them. We generally patrol a site three times in the summer as they do not all flower at the same time. So, we spray once in early July, early August, and early September. (the two loosestrife eating insects are also an option. I am personally opposed to introducing another exotic organism to our systems, but many in TNC think it is the only option. I can send you a contact if you like).

In general, I have found fens to be tricky places to use herbicide - especially foliar application. Grasses and sedges are particularly sensitive and you can get large brown-outs if you are not careful. As I said above, dense stands of Phragmites are better targets for foliar spray as the overspray is less important. If the Phragmites is intermingled with native species, and you can get the help, hand-cutting may be the way to go. I think you have to bite the bullet with purple loosestrife and accept a small amount of overspray in order to control it. We have found that oversprayed areas are re-colonized by natives pretty quickly. As a colleague always reminds me, doing nothing is a management strategy also - in the case of purple loosestrife means the end of your natural area - and this may be the most important thing you can impress on folks.

I have tried to locate info on herbicides and turtles with no luck. A couple of ideas that you may have already thought of : (1) does FWS know where the preferred nesting grounds are? These are often the high and dry parts of the fen complex and can be avoided, (2) These species are usually in low densities, so your chance of spraying individuals directly is very low, (3) I have yet to encounter a turtle in a stand of Phragmites - it is too difficult to get into.

Chip O'Leary
Restoration Coordinator, TNC - Indiana Chapter

IIB. Case study: Control efforts in Indiana

Here in Indiana, I manage several fens with invasive plant problems. I have never read anything that said glyphosate was bad for turtles. The story is that the surfactant used in RoundUp (glyphosate) is bad for amphibians and other wetland animals, so that is why Rodeo (glyphosate) is used, which is basically RoundUp without the surfactant.

We have been spraying Phragmites australis in the early fall, September through October, when the seed head is fully emerged, with Rodeo at recommended rates with a solo backpack sprayer with a 5-foot wand. If you spray the top leaves of the plant they die really easily within a few weeks and then one moderate follow-up treatment is needed the next year. If you are careful, you rarely get any herbicide on other plants. We treated areas up to 5-6 acres in size and have had great success. The next year native plants come back from the seed bank under the mat of dead vegetation. A follow-up prescribed burn can also help to remove the large mat of dead Phragmites.

Sometimes for Phragmites, we cut the plant a few inches above the ground and drip herbicide into the cut stem at about 25%, and that has about 50% success, and this is helpful in areas you do not want to overspray or are sensitive. I think herbicide is the best control method for Phragmites. It works well and overspray can easily be minimized in thick stands.

For purple loosestrife (Lythrum salicaria) we have been spraying in a similar way when the flowers appear on the plant. Herbiciding is done frequently throughout the late summer and early fall, especially on flowering plants. It will take several years of spraying to make a real impact on the site. If you are careful and use an adjustable spray nozzle you can really minimize overspray. It is tough when you see some browning around the area of natives, but just remember they are generally tougher than exotics, and will come back soon! You can also replant the area with native plugs and that helps out a lot. Loosestrife is tough and there is generally some overspray, but native plants quickly come back. It has a large seedbank and takes many years to deplete. I would not replant a loosestrife area with natives for awhile, since the seedbank can take several year to deplete.

Some DNR people in Indiana have released the beetle that eats loosestrife, and have seen pretty good success so far. The beetle may help deplete the seed bank quicker than herbiciding. Our herbicide work on TNC property for loosestrife is coming along really well after only two years of spraying.

Mike Norris, Restoration Specialist, Southern Lake Michigan Rim Project

IIC. Case study: Control efforts in New Jersey

What methods have you used that have been effective in controlling the weeds, but don't negatively impact the native vegetation?

In northern New Jersey, we have used cut-stem treatment to control both Phragmites and purple loosestrife at several wetland sites. We have found that injecting the cut stems of Phragmites with Rodeo (50%) after the tassel stage (mid-September-October) is very effective. We thought of removing flowering spikes from the site but since staff was limited, we did not follow through with this idea. Later we learned that seeds are often not viable and germination and successful seedling establishment is rare, so we thought it was not worthwhile to remove the flowers.

The cut-stem treatment appears to have killed most of the underground root system because there has been little to no re-sprouting 2 years after control. While this method results in minimal off-target damage, it is very labor intensive. It will take us three years to treat several small infestations of Phragmites on one of our preserves using both volunteers and 4 full-time staff. This method is probably not worthwhile for people who have large infestations.

This year we also began controlling purple loosestrife in an emergent marsh using cut-stem treatment. Stems were cut near the base and Rodeo (30%) was squirted onto cut stems when plants began flowering (July-August). Herbicide was applied only to stems of target plants, thereby reducing exposure to other species. Cut plant material was bagged and removed from the site. Since control was initiated this year, we do not have information about the effectiveness of this method yet.

What is the most effective means of controlling the weeds (manual, pesticide, combination)?

We have found a combination of manual removal (i.e. cutting) followed by pesticide application to be most effective. See answer to #2.

If you're herbiciding, have you done your treatments stepwise or have you done the herbiciding all at once? It would be useful to hear how large the areas are that you are treating.

Whenever possible we have treated the entire infestation at once. In situations where we don't have enough time or staff to treat the whole area we have been working on patches of the infestation in a stepwise manner.

The Phragmites infestations vary in size. We have treated small patches with only several dozen stems to patches 50 X 25 m. This fall we plan to use volunteers to help us finish treating a Phragmites infestation that is approximately 40 X 40 m. Most of the Phragmites infestations are dense monocultures with only a few other native species present.

The area where we treated purple loosestrife was approximately 0.5 acres. However, this area had scattered clumps of purple loosestrife throughout (approximately 250 stems).

Finally, have you followed up with any planting of native plants or have they filled in the blank spots left when the weeds are killed? This ties in with question #4.

To date (1-2 years after treatment) only a handful natives have moved in where the dense Phragmites clones once stood. It appears that it will take sometime before natives fully repopulate the area.

Melissa Morris, Science and Stewardship Assistant, TNC-New Jersey

IID. Case study: Control efforts in Washington State

I've been involved with purple loosestrife (Lythrum salicaria) control for about three years on some private property here in Washington State, and the most effective method we've found is winter pulling. We have not tried any herbicides, but have done summer pulls and found winter pulls to be much more effective because: 1) the area is much wetter and plants uproot more easily and completely; 2) less biomass has to be removed and disposed of; and 3) the area can be revisited the following summer for removal of root remains that have re-sprouted. Here, the wetland freezes only sporadically during the winter, and we make a Christmas-time party out of it to get volunteers. Incidentally, we have also tried release of loosestrife-eating beetles (Galerucella pusilla, G. calmariensis), for bio-control, but although I've heard good reports about them in other areas, they didn't seem to have been able to persist after the first release on our site.

Questions:

How do you uproot these monsters...just grab and pull...since all that is left in the winter are the brown dead stalks? Do you use a spade? Is there much soil disturbance with this method?

Indeed, it's grab and pull. I've found that this is much harder in the summer, and you have to use a spade, but here the area is wet enough in winter, and there's enough of a stalk left, that a good, careful pull gets almost all of the root-ball out. The few bits that break off you can catch the next summer when they reveal themselves by re-sprouting. The grab and pull is actually quite satisfying! And since it's so wet, it's much easier to get the excess soil off. I've done this for two winters now and will never go back to summer pulls or cuts (tried cuts too and found them much less effective).

Are the old seed heads bagged first? Or have the dead seed heads already dispersed their seeds?

One winter, we pulled in the same place where we had earlier cut seed heads, so they were already gone. The next year (this last one), we did not deal with the seed heads separately at all. I suppose to be most thorough, this would be a good idea, but I've found the winter pull (followed by summer touch-up) so much more effective than anything else that we didn't put extra energy into bagging seed heads, reckoning we'll be monitoring this situation for many years to come and will catch seeds if they get far enough to come up. (Also, there are nearby loosestrife areas not being controlled, and they'll be producing plenty of seed we can't control)

Is there a lot of re-sprouting the next year? Can you actually eradicate this species after a number of years?

In two years (July 98-now) in the area where we've been doing this method, we've pretty much gotten rid of the loosestrife, although as I said I'm sure there will be a need for ongoing monitoring of stray seed- and root-sproutings. The loosestrife in this area when we started was established and rapidly-expanding, but not yet an uninterrupted mat; now it's open water, rushes and other emergents again. This is in contrast to adjacent areas where there's been some cutting of seed heads and some no-control-at-all, where the loosestrife is continuing its relentless takeover. I plan to try another beetle release in some of these other areas, where the loosestrife mat is very dense, and continue to use the winter-pull method to reduce infested areas at the edges and stop any new invasions.

How do you keep volunteers out in the cold water? Is there any danger of hypothermia? Do you wear wetsuits (or drysuits)?

Yes, the cold is something to consider. We were thigh-high in water, and simply made sure there were plenty of warm dry clothes and hot cocoa waiting--but it would probably be better to use wetsuits or waders. The only other factor to consider is that the bottom can be quite mucky and my boots got stuck several times before I decided to take them off. But it was only in the low-50's when we attempted the winter-pulls . It worked well to have only a few people actually that deep in the water, and several others wearing knee-boots at the edges, catching the plants the others pulled from the deep. You can make a great game of it--the root-ball makes a weighty mass at one end and the stalk is the handle--throw it javelin-style, or twirl it and let it fly in the direction of those waiting! (As you can see, we actually had fun with this, and got the loosestrife out to boot!)

Marcy Summers, The Nature Conservancy of Washington

IIIA. Methodology: Applications of herbicide (Michigan)

One of our fen sites has a federally-endangered butterfly, so we have had to evaluate the impacts of herbicide and trampling. We use ACCORD, which is glyphosate and wetland approved, and half the price of RODEO. Our methods have been simplified so that volunteers can do the work without a lot of thought process.

For both loosestrife (Lythrum salicaria) and Phragmites, we use a an active mix of 5% or less (ACCORD is 41% active in concentrate and we dilute 1 part concentrate to 7 parts water). Each plant is cut and treated. Loosestrife is cut "high" (just below inflorescence) so that plant will keep growing and absorb herbicide. If cut too low, the individual "gives up" on the leader and re-sprouts from the roots. For Phragmites all stems are cut low (even if they appear dead) and treated. We use a PVC applicator with sponge tip (design available from my MI office, contact Sara Ross, or off-of Intranet tools of the trade). The loosestrife is wiped up the stem and then the cut is also treated; only the cut is treated on the Phragmites., but more herbicide is used on Phragmites so that the strawlike structure absorbs the mix.

We also use this PVC applicator and ACCORD on woody plants, 14% active in mix (1:2) with great success. Our treatment is in patches and stepwise throughout growing season, but most intense when loosestrife is blooming (easier to locate), and in late July/August for Phragmites. To reduce trampling we try to travel through the densest patches as we treat, and have a 1 or two person team for the isolated bunches.

As for glyphosate and herps...we try to avoid dripping any mix into water, and use the wipe method since we can more carefully control the herbicide. We do not spray unless we are in solid patches, and that is on a case-by-case decision.

Jack McGowan-Stinski, Michigan Chapter, The Nature Conservancy

We do some control with Rodeo (glyphosate) - using what we call the Bloody Glove method, which we got from Jack McGowan-Stinski at the Michigan chapter. We are not dealing with any rare herps, but we're still looking for the best strategy for controlling the loosestrife. We do the herbiciding immediately after snapping off the flower heads, but we only work in the highest quality sites where the infestations are small.

Dave Borneman, Coordinator, Ann Arbor Department of Parks and Recreation

The Bloody Glove technique

We use a 5% solution of Rodeo (glyphosate; ai = 53.8%) in a spray bottle, which we add dye to. The herbicider wears chemical resistant nitrile or latex gloves on both hands covered by a fleecy, cotton glove on one of the hands. The herbicider follows behind the person removing and bagging the flower heads. Grab the loosestrife by the hand with the cotton glove and spray the herbicide into that hand (trying not to hit non-target plants). Then wick the loosestrife up the top 1/3 of the plant. As the cotton glove becomes saturated with herbicide, you can spray less often. After herbiciding, we put the cotton gloves in a plastic container and leave the lid cracked. (If you put the lid all the way on, it can become moldy.) We can usually re-use the cotton ("bloody") gloves for quite awhile. And of course you can just rinse off the nitrile or latex gloves.

The most challenging part of the herbiciding process is not getting the herbicide all over you. Sometimes it can get tricky to tell where you have herbicided and you find yourself walking through herbicide coated loosestrife. Because of this, we require our crew to wear chemical resistant Tyvek when controlling loosestrife since it can get messy. That aside, we have found this technique to be very effective.

Kristin Condict, Technician, Ann Arbor Department of Parks and Recreation

IIIB. Adaptive management for weed control

I am working on a similar problem in an inland salt marsh, which has some obviously different properties from a fen. Below, see the adaptive management approach we have been using for the last 5 years, and are seriously adapting this year.

The repeated mowing had reduced the vigor of the Phragmites stand slightly but was adding lots of organic matter to the system, lots of trampling and really was not accomplishing what we needed it to. We are not managing purple loosestrife in the system, which is abundant but does not infiltrate the core salt marsh area. Beetles have been introduced in the area by USFWS, however, so we will see about that...

Bill Patterson, Field Representative, TNC - Central/Western New York Chapter

Carncross Preserve Project Plan: Developing Capacity for Invasive Plant Monitoring and Control

Project Plan
Carncross Preserve contains a globally rare, inland salt marsh community unique in its status as the highest quality example the community type in the Montezuma Wetlands complex and New York State. Distinct in species composition and ecological function from coastal salt marsh systems, inland salt marsh communities are formed by localized underlying salt deposits. Acquisition at Carncross of 100 acres of salt marsh habitat and buffer by The Nature Conservancy (TNC) in the 1980's has protected the site from salt mining, agriculture, and hydrologic alteration. However, the viability of the site remains threatened by the presence of invasive species including purple loosestrife and phragmites. Purple loosestrife, with it's low salt tolerance, represents only a mild threat to the core salt marsh area, yet the highly invasive and salt tolerant phragmites clones present at the site threaten to obliterate the salt marsh community if left unmanaged.

To address the phragmites problem at Carncross, TNC staff and volunteers began mowing the phragmites in 1995 and laid out 3 twenty-meter transects to evaluate the results. While mowing has visually reduced the vigor of phragmites, no advance or retreat of the phragmites is observed due to mowing. Monitoring of transects confirms this observation. Based on information available in The Nature Conservancy's Element Stewardship Abstracts, herbicidal control of phragmites is highly effective and can be implemented with minimal residual damage.

Management Objective
: Restoration and maintain the native inland salt marsh plant community at Carncross salt pond to 100% of potential area by eliminating (0% cover) invasive phragmites clones by spring 2003.

Monitoring Objectives:
1. Through the use of existing aerial photos and site visits, determine the extent of the potential and existing inland salt marsh community as a target for restoration.
2. Track effectiveness of herbicide applications over mud flats through the use of transects to locate the position of three representative phragmites clones.
3. In 2000 evaluate three separate methods for herbicide treatment of phragmites over native salt marsh plants while minimizing residual damage.

Note: Similar transects with photos were completed in 1995, 1996, 1997, 1998, and 1999. Data from the transect are summarized in Jim Marks' report dated November 13, 1998. Various sequences of photos from these years can be found in the Carncross files.

1. Aerial Photos- Aerial photos from 1999, 1989, and earlier if available to will be analyzed to map the current and potential extent of inland salt marsh community potential habitat. Location of phragmites clones will be overlaid to show extent of invasions. Identification of boundaries will occur on the ground and be related to aerial photos by GIS digitizing if available. Inland salt marsh community will be defined as the presence of a suite of inland salt marsh community indicator species such as Spartina sanisarodies, Hordeum jubatum, agropyron repens, quenapodium sp., Scirpus americanus, Scirpus meridamus, among others and is generally found between a lower mud flat boundary and an upper boundary defined by the salt intolerant.

Return to List




## Japanese stiltgrass, Nepalese browntop
## Microstegium vimineum (Trin.) Camus

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is September, 2004.

**NATIVE RANGE**: Japan, Korea, China, Malaysia and India

**DESCRIPTION**: Japanese stilt grass, also known as Nepalese browntop, is an annual plant in the grass family (Poaceae).




Photo: Chris Evans, The Univ. of Georgia, www.invasive.org.

It has a sprawling habit and grows slowly through the summer months, ultimately reaching heights of 2 to 3 1/2 ft. (6-10 dm.).




Photo: James H. Miller, USDA Forest Service, www.invasive.org

The leaves are pale green, lance-shaped, asymmetrical, 1-3 in. (3-8 cm.) long, and have a distinctive shiny midrib.




Photo: David J. Moorhead, The Univ. of Georgia, www.invasive.org.

Slender stalks of tiny flowers are produced in late summer (August - September). The fruits or achenes mature soon after flowering and the plant dies back completely by late fall.

**ECOLOGICAL THREAT**: Japanese stilt grass is especially well adapted to low light conditions. It threatens native plants and natural habitats in open to shady, and moist to dry locations.




Photo: Chris Evans, The Univ. of Georgia, www.invasive.org.

Stilt grass spreads to form extensive patches, displacing native species that are not able to compete with it. Where white-tail deer are over-abundant, they may facilitate its invasion by feeding on native plant species and avoiding stilt grass.

**DISTRIBUTION IN THE UNITED STATES**: Japanese stilt grass has been reported to be invasive in natural areas in fourteen eastern states (Connecticut, Delaware, Georgia, Indiana, Kentucky, Maryland, North Carolina, New Jersey, New York, Pennsylvania, Tennessee, Virginia, Wisconsin, West Virginia) and Washington, D.C. Click here to see a distribution map.

**HABITAT IN THE UNITED STATES**: Stilt grass occurs in a wide variety of habitats including moist ground of open woods, floodplain forests, wetlands, uplands, fields, thickets, paths, clearings, roadsides, ditches, utility corridors, and gardens. It readily invades areas subject to regular mowing, tilling, foot traffic, and other soil disturbing activities. Stilt grass appears to prefer moist, acidic to neutral soils that are high in nitrogen.

**BACKGROUND**: Introduced into Tennessee around 1919, stilt grass may have accidentally escaped as a result of its use as a packing material for porcelain.

**BIOLOGY & SPREAD**: Japanese stilt grass is a colonial species that spreads by rooting at stem nodes that touch the ground. Stilt grass reproduces exclusively by seed. Individual plants may produce 100 to 1,000 seeds that fall close to the parent plant. Seed may be carried further by water currents during heavy rains or moved in contaminated hay, soil, or potted plants, and on footwear. Stilt grass seed remains viable in the soil for five or more years and germinates readily.

**For more information on the management of Japanese stilt grass, please contact:**

Art Gover, Penn State University, LMRC, Orchard Rd., University Park, PA 16802; (814) 863-1184; aeg2@psu.edu.
Fred Yelverton, Box 7620, NCSU Campus Raleigh, NC 27695-7620, (919) 515-5639 (Fred_Yelverton@ncsu.edu).
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Nonnative Invasive Plants of Southern Forests, http://www.invasive.org/eastern/srs/
Southeast Exotic Pest Plant Council - Invasive Plant Manual. www.se-eppc.org.
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html

Virginia Natural Heritage Program Fact Sheet-Japanese stiltgrass; http://www.dcr.virginia.gov/dnh/invlist.htm

**AUTHOR**: Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**REVIEWERS**: Nancy Benton, Gwendolyn Thunhorst.

**REFERENCES:**

Barden, Lawrence. 1987. Invasion of Microstegium vimineum (Poaceae), an exotic, annual, shade-tolerant, C-4 grass, into a North Carolina floodplain. The American Midland Naturalist 118 (1):40-45.
Barden, Lawrence. 1991. Element Stewardship Abstract: Microstegium vimineum. The Nature Conservancy.
Fairbrothers, D. E. and J.R. Gray. 1972. Microstegium vimineum (Trin.) A. Camus (Gramineae) in the United States. Bulletin of the Torrey Botanical Club 99:97-100.
Hunt, D. M. and Robert E. Zaremba. 1992. The northeastward spread of Microstegium vimineum (Poaceae) into New York and adjacent states. Rhodora 94:167- 170.
LaFleur, A. 1996. Invasive plant information sheet: Japanese stilt grass. The Nature Conservancy, Connecticut Chapter Connecticut, Hartford, CT.
Miller, J.H. 2003. Nonnative invasive plants of southern forests: a field guide for identification and control. Gen. Tech. Rep. SRS-62. Asheville, NC: U.S. Department of Agriculture, Forest Service, Southern Research Station. 93 pp.
Redman, Donnell E. 1995. Distribution and habitat types for Nepal microstegium [Microstegium vimineum (Trin.) Camus] in Maryland and the District of Columbia. Castanea 60(3): 270-275.
Rhoads, A.F. and T.A Block. 2000.The Plants of Pennsylvania, An Illustrated Manual. University of Pennsylvania Press. 1061 pp.
Swearingen, J. 2004. WeedUS: Database of Invasive Plants of Natural Areas in the U.S. Plant Conservation Alliance. http://www.nps.gov/plants/alien
USDA, ARS, National Genetic Resources Program. Germplasm Resources Information Network - (GRIN) [Online Database]. National Germplasm Resources Laboratory, Beltsville, Maryland. URL: http://www.ars-grin.gov/var/apache/cgi-bin/npgs/html/taxon.pl?2964 (01 September 2004).

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Japanese stiltgrass, Nepalese browntop
# *Microstegium vimineum* (Trin.) A.Camus

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese stiltgrass- http://www.nps.gov/plants/alien/fact/mivi1.htm - Author: Jil Swearingen - September 10, 2004**

A variety of control methods are available for stilt grass, depending on the extent of the infestation, the type of habitat, and the availability of labor and other resources. Preventing the introduction of stilt grass into non-infested areas and out of infested areas should be a priority. Early control of new infestations will also reduce the likelihood of establishment.

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

Potential for Restoration of Invaded Sites: Manual and mechanical, environmental/cultural, and chemical methods are all useful to varying degrees in controlling M. vimineum. Prescribed burns have not been successful in controlling this species so far, but fall burns may have the potential for partial control. M. vimineum produces a large number of viable seed that can remain in the soil seed bank for seven years or more. If controlled during the early stages of invasion, the potential for successful management is high. The potential for large-scale restoration of wildlands where M. vimineum has become established is probably moderate.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese stiltgrass- http://www.nps.gov/plants/alien/fact/mivi1.htm - Author: Jil Swearingen - September 10, 2004**

Manual: Stilt grass is a shallow-rooted annual that can be pulled by hand throughout the growing season, especially when the soil is moist and entire plants with roots can be removed. Pulling is easier and probably more effective in mid-to-late summer when the plants are much taller and more branched. At this stage, entire plants can be easily removed by grabbing the basal portion of a plant and pulling firmly. In short time, a fair amount of stilt grass can be pulled and piled up to dehydrate on site. If plants are already in the fruiting stage, they should be bagged and disposed of offsite to prevent dispersal of seed. Also, try to avoid pulling native grasses like Virginia cutgrass (Leersia virginia) that often grow intermingled with stilt grass and may be difficult to distinguish from it. Because hand pulling plants disturbs the soil and may expose stilt grass seed from previous seasons, late season pulling will avoid the likelihood of seed germination. Hand pulling of plants will need to repeated and continued for many seasons until the seed bank is exhausted.

Mechanical: Stilt grass can be mowed in late summer (i.e., August through September) when the plants are flowering but preferably before seed is produced. This can be done using a lawn mower or "Weed Whacker" type machine or a scythe. Because stilt grass is primarily an annual plant, cutting late in the season before the plants would die back naturally avoids the possibility of regrowth. Recent information suggests that stilt grass plants cut early in the summer respond by and regrowing and flowering soon after cutting, much earlier than they would normally flower. Another reason to cut late in late summer to fall.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Bush honeysuckle - http://www.se-eppc.org/manual/japgrass.html -2003**

Mow plants as close to the ground as possible using a weedeater or similar grass cutting tool. Treatments should be made when plants are in flower and before seeds are produced. Treatments made earlier may result in plants producing new seed heads in the axils of lower leaves.

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

Manual Control: Hand pulling of M. vimineum is the preferred method of removal as it is highly specific and provides minimal impact (except trampling and soil disturbance) to the surrounding environment. Hand pulling is an effective method of control if it is thorough and timed correctly. It is, however, labor-intensive and time-consuming. Pulling late in the season (September-early November) before seed production reduces the unintentional spread of the current year's seeds. Pulling early in the season (before July), however, allows germination of new plants from the seed bank which will mature during the remaining season and produce seeds. In the northeast, August and late September are good times to pull plants by hand (LaFleur 1996). Yearly weeding is necessary because new plants can appear as a result of seed banking or re-infestation from new seed being dispersed into the area (G. Edinger, letter to J. Randall).

Mechanical Control: Mowing using a weed whacker (or a weed-eater) is an effective control method if carried out in late summer just before seeds are produced. Mowing at any other time is not useful as the plants have the ability to resprout and can produce seed heads in the axils of their lower leaves (Woods 1989; Barden 1991). Mowing can also be useful in reducing the amount of litter and plant biomass prior to herbicide application, making the herbicide more effective.

Flooding: Flooding for more than three months, or intermittent flooding during the growing season, may be an effective control method for mature plants of M. vimineum. The seeds of M. vimineum, however, can survive periods of inundation of at least ten weeks (Barden 1991).

## GRAZING:

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

Grazing is not a control option for M. vimineum since cattle, deer, and even goats avoid feeding on it (A. Houston, pers. comm.; Barden 1991).

## PRESCRIBED FIRE:

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

Spring burns are ineffective at controlling M. vimineum because a new cohort of seeds will germinate soon after the burn. Burns in the late fall, however, may be useful in controlling this species (Barden 1991). Burning is also useful in reducing the amount of litter and plant biomass prior to herbicide applications.

## BIOCONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese stiltgrass- http://www.nps.gov/plants/alien/fact/mivi1.htm - Author: Jil Swearingen - September 10, 2004**

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

No biological controls are currently available for this plant.

## CHEMICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese stiltgrass- http://www.nps.gov/plants/alien/fact/mivi1.htm - Author: Jil Swearingen - September 10, 2004**

For extensive stilt grass infestations, use of a systemic herbicide such as glyphosate (e.g., Roundup) is a more practical and effective method. If applying glyphosate to stilt grass in wetland sites, use the formulation labeled for wetland areas (e.g., Rodeo). Apply a 2% solution mixed with water (8 oz. per 3 gals. mix) and a surfactant in late summer. Be careful to avoid application to non-target plants because glyphosate is a non-specific herbicide that will kill or damage most plant species it contacts.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Bush honeysuckle - http://www.se-eppc.org/manual/japgrass.html -2003**

Herbicide treatments should be made late in the growing season but, before the plants set seed. Treatments made earlier in the growing season may allow a second cohort of plants to produce seeds.

> Glyphosate: Apply a 2% solution of glyphosate and water plus a 0.5% non-ionic surfactant to thoroughly wet all foliage. Do not spray to the point of runoff. Ambient air temperature should be above 65°F to ensure translocation of the herbicide to the roots. Do not apply if rainfall is expected within two hours following application.

Sethoxydin: Apply a 1.5% solution of sethoxydin and water plus a 1% nonphytotoxic vegetable-based oil to all foliage on a spray-to-wet basis. Do not spray to the point of runoff. Ambient air temperature should be above 65°F. Do not apply if rainfall is expected within one hour following application.

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

For large infestations of M. vimineum, the use of herbicides may be the only viable option for good control. A series of control experiments using herbicides was carried out at the Ames Plantation (University of Tennessee), and the researchers reported that it is relatively easy to kill M. vimineum, but that managing for a desirable plant community is difficult. They found that the herbicide imazameth (tradename Plateau) was the herbicide of choice for controlling M. vimineum. This is because imazameth (applied at a rate of 6 ounces per acre) kills M. vimineum, but allows the development of (a.k.a., does not kill) the desirable native sedges, ragweeds, and legumes.

The grass-specific herbicide fluazifop-p (tradename Fusilade) applied at the rate of 1.2 liters per hectare (1 pint per acre) also controlled M. vimineum, but left a less desirable plant community. Glyphosate (tradename RoundUp) was also tested, but resulted in a complete kill of all plants, which could potentially lead to possible re-invasion by M. vimineum or other undesirable species. Barden (1991) also found glyphosate useful in killing M. vimineum. Formulations of glyphosate registered for use aquatic systems (Rodeo), has been effective for M. vimineum control in wetlands. Woods (1989) in Tennessee found that the grass-specific herbicide sethoxydim (tradenames Poast, Vantage), applied during late summer at rates of 1 pint per acre, also provided excellent (more than 95%) control of M. vimineum and released dicots from competition without injuring them. Pre-emergent herbicides such as diphenamid (tradename Enide) and benefin (tradename Balan) have also demonstrated excellent control of M. viminium seedlings under conditions of good herbicide-to-soil contact (Woods 1989), but do not encourage the germination of native species.

Allan Houston (pers. comm.) reports that if there is a heavy build-up of litter (dead plant material) in M. vimineum stands, burning the debris may first be necessary to get adequate herbicide coverage. He suggests applying herbicide when the plants reach a height of 5-10 centimeters (2-4 inches).

**Nonnative Invasive Plants of Southern Forests: a field guide for identification and Control: Bush honeysuckles - Gen. Tech. Rep. SRS-62. Asheville, NC, U.S. Department of Agriculture, Forest Service Research Station - http://www.invasive.org/eastern/srs/NB.html - Author: James H. Miller - 2003**

Recommended control procedures:

- Apply a glyphosate herbicide as a 2-percent solution in water (8 ounces per 3-gallon mix) with a surfactant in late summer. Or, apply Vantage (see label) for situations that require more selective control and less impact on associated plants.
- Repeat treatments for several years to control abundant germinating seeds. Mowing or pulling just before seed set in September will prevent seed buildup.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Species Management Summary (ESA): Japanese stiltgrass - http://tncweeds.ucdavis.edu/esadocs/micrvimi.html - Author: Mandy Tu - August 2000**

EXAMPLES OF MICROSTEGIUM VIMINEUM MANAGEMENT ON TNC PRESERVES:

According to TNC's 1998 Weed Survey, M. vimineum has been reported from TNC preserves in New Jersey, North Carolina, Virginia, Georgia, Alabama, Arkansas, Maryland, and in Connecticut. Several preserves reported M. vimineum is one of their worst weed problems, but only a few had begun active control measures.

In Maryland, Donnelle Keech reported that burning is not effective in controlling M. vimineum. In North Carolina, Robert Merriam reported hand pulling was effective. Elizabeth Farnsworth in Connecticut, however, indicated that hand pulling may be effective in small populations, but seems futile for large populations since it is difficult to eliminate the seed sources. She added that it is important to attack small infestations as soon as possible, and to attack them vigorously!

CONTACTS:

Elizabeth Farnsworth or David Gumbart
The Nature Conservancy
Connecticut Chapter
55 High Street Middletown, CT 06459
(860) 344-0716
efarnsworth@tnc.org

Doug Samson
The Nature Conservancy
Maryland/DC Chapter
2 Wisconsin Circle, #300
Chevy Chase, MD 20815
(301) 656-8673

Robert Merriam

4011 University Drive
Durham, NC 27707
(919) 403-8558
bmerriam@tnc.org

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Star-of-Bethlehem
## *Ornithogalum nutans* L. and *Ornithogalum umbellatum* L.

**SOURCE**: The following information on this species is taken from the Invasive Plant Atlas of New England (IPANE) website at http://invasives.eeb.uconn.edu/ipane/ and Rhoads and Blocks, The Plants of Pennsylvania: An Illustrated Manual. Modifications include additional resource information. Check the website links for future updates.

**DESCRIPTION:** *Ornithogalum umbellatum* is an herbaceous plant that grows from bulbs that are renewed each year. The bulbs are ovoid in shape and measure (0.5-1.5in.) in length. The coat of the bulb is membranous. The leaves of *O. umbellatum* are narrow and linear, measuring 10-30cm (4-12in.) long and 2-5mm (0.1-0.2in.) wide. The leaves form a tuft from which the scape emerges. The leaves of O. nutans are 8 - 15 mm wide.

The white, star-shaped flowers are borne on a scape that can grow up to 30cm (1ft.) in height, but is usually shorter. The umbel-like raceme contains 3-10 flowers. Each flower has 6 petals and measures 1.5-2cm (0.5-0.75in.) in diameter and1- 2 cm. in length. *O. nutans* flowers measure 2-2.5 cm in length. The lower pedicels, or flower stalks, of *O. umbellatum* are >1cm and longer than the bracts, whereas, they are <1 cm. and shorter than the bracts in *O. nutans*. The outside of the flower in both species is green and white margined. The flowers appear on the plant from May to June. The capsules of this plant are subglobose in shape and 3-sided. Each locule contains many black seeds.

Page References Bailey 232, Fernald 437, Gleason & Cronquist 829, Holmgren 787, Magee & Ahles 348, Newcomb 334. See reference section below for full citations. Rhoades and Block. 2000. The Plants of Pennsylvania: An Illustrated Manual. University of Pennsylvania Press. 1061 pp.



Photos: Leslie J. Mehrhoff (top 2 pix) and Stacey Leicht (bottom), University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/

**SIMILAR SPECIES:** None

**REPRODUCTIVE/DISPERSAL MECHANISMS:** *Ornithogalum umbellatum* disperses itself by means of its bulbs, which can be dispersed by water. The extent to which it is able to reproduce by seed in the New England region is unclear. (We will provide further information on this question as it becomes available to us.)

**DISTRIBUTION:** *Ornithogalum umbellatum* is native to North Africa as well as Europe. In Europe, it is found from Portugal and Spain in the west, south to Italy, north to parts of France and east to Turkey. In the United States, this plant is found from Maine to Florida including all the states west to South Dakota, Nebraska, Kansas, Oklahoma and Texas. It is also found in Washington, Oregon, Idaho and Utah. This plant has been reported from all New England states.

**HISTORY OF INTRODUCTION IN NEW ENGLAND:** Due to its attractive flowers, *Ornithogalum umbellatum* was most likely introduced into New England as a horticultural plant. The exact date of introduction is not known, but there were reports of large stands of it in 1940 in the forests of Indiana. It was most likely dispersed via additional plantings as well as the bulbs being washed downstream.

**HABITATS (IN NEW ENGLAND)**: Early Successional Forest, Edge, Floodplain Forest, Wet Meadow, Yard or Garden

This plant prefers moist to wet habitats. It is most often seen along the banks of rivers and streams in disturbed situations.

**THREATS:** Since this plant is still being used horticulturally, it is likely to continue spreading beyond its current range. *Ornithogalum umbellatum* is poisonous if not lethal to livestock and should be prevented from spreading into agricultural situations. *Ornithogalum umbellatum* produces new bulbs each year, and these bulbs are readily moved downstream to new localities. It has the ability to form locally dense stands along the edges of the rivers and streams, allowing it to crowd out native riparian plants.

**For more information on Star-of-Bethlehem, contact:**
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA - NRCS PLANTS Database, http://plants.usda.gov/

**REFERENCES:**
Bailey, L. H. 1949. Manual of Cultivated Plants. Macmillan, New York.
DeMars, B.G. 1994. Star-of-Bethlehem, Ornithogalum umbellatum L. (Liliaceae): An invasive, naturalized plant in woodlands of Ohio. Natural Areas Journal 14(4): 306-307.
Fernald, M.L. 1950. Gray's Manual of Botany 8th ed. American Book Co., Boston.
Gleason, H. A. 1952. The New Britton and Brown Illustrated Flora of the Northeastern United States and Adjacent Canada. Macmillan Publishing Co., Inc. New York
Gleason H.A. and A.C. Cronquist. 1991. Manual of Vascular Plants of the Northeastern United States and Adjacent Canada. 2nd ed. New York Botanical Garden, Bronx, New York.
Haragan, P.D. (1991). Weeds of Kentucky and the Adjacent States, A Field Guide. The University Press of Kentucky.
Holm, L.G., Pancho, J.V., Herberger, J.P., and Plucknett, D.L. (1979). A Geographical Atlas of World Weeds. John Wiley and Sons, New York, USA.
Holmgren N.H. 1998. Illustrated Companion to Gleason and Cronquist_s Manual. New York Botanical Garden, Bronx, New York.
Magee D.W and H.E. Ahles. 1999. Flora of the Northeast. University of Massachusetts Press, Amherst.
Mezev-Krichfalishii, G.N., Krichfalushii, V.V., and Komendar, V.I., (1989). Studies on the population biology of Ornithogalum umbellatum L. (Liliaceae) for elaborating a strategy of species survival in Transcarpathia. Tiscia 24,3-10.
Moret, J. (1992). Numerical taxonomy applied to a study of some ploidy levels within the Ornithogalum umbellatum complex (Hyacinthaceae) in France. Nordic Journal of Botany 12, 183-197.
Murphy, T.R. and Johnson, B.J. Weeds of Southern Turfgrasses. The College of Agricultural and Environmental Sciences, Griffin, Georgia.
Newcomb N. 1977. Newcomb_s Wildflower Guide. Little Brown, Boston.
Rhoades and Block. 2000. The Plants of Pennsylvania: An Illustrated Manual. University of Pennsylvania Press. 1061 pp.
USDA, NRCS. 2001. The PLANTS Database, Version 3.1. (http://plants.usda.gov). National Plant Data Center, Baton Rouge, LA 70874-4490 USA.
Uva, R.H., Neal, J.C, and DiTomaso, J.M. (1997). Weeds of the Northeast. Cornell University Press.

**Management and Control Information**

Return to List




## Species Management and Control Information

### Star-of-Bethlehem
### *Ornithogalum nutans* L. and *Ornithogalum umbellatum* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

**Management information for this species was unavailable at time of tutorial development**

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Wild Parsnip
## *Pastinaca sativa* L.

**SOURCE**: The following information on this species is taken, in part, from the Illinois Nature Preserves Commission, Illinois Vegetation Management Guidelines for Wild Parsnip, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html. . Modifications include additional pictures from www.invasives.org and http://tncweeds.ucdavis.edu/photos.html. Check the website links for future updates. The last update for this information is February, 1990.

**DESCRIPTION:** The thick taproot of the wild parsnip is long, conic, and fleshy. Branching from the fleshy root is the light green, hollow, deeply-grooved stem that stands erect at 2-5 feet (0.6-1.5 meters) tall. Leaves are alternate, pinnately compound, and branched with saw-toothed edges. Each leaf has 5-15 ovate to oblong leaflets with variably toothed edges and deep lobes. The petiolate lower leaves are often 1.5 inches (3.8 cm) long, while upper leaves are sessile and much reduced. The small, 5-petaled, yellow flowers are arranged in 2-6 inch (5-15 cm) broad umbels at the top of slender stems and branches. Each compound flat umbel has 15-25 primary rays that contain yellow blossoms during the June-September flowering season. The blossoms give rise to a fruit termed a schizocarp that is broadly oval and 0.25 inch (6 mm) long. The abundant 0.25 inch (6 mm) mericarps (segments of the fruit) of this parsnip are flat, round, smooth, straw-colored, and have low ribs across them.








Photos: Upper right: Chris Evans, University of Georgia, www.forestryimages.org

Others: © John M. Randall/The Nature Conservancy

**SIMILAR SPECIES:** Wild parsnip is distiguished from other species in the parsley family by its yellow flowers and its pinnately compound leaves that are divided once into more than five leaflets. Wild parsnip should be accurately identified before attempting any control measures. If identification of wild parsnip is in doubt, the plant's identity should be confirmed by a knowledgeable individual and/or by consulting appropriate books.

**DISTRIBUTION:** This member of the parsnip or umbel family has escaped from cultivation and is common throughout the northern United States and Canada, from British Columbia to California and Vermont, and south to Florida. In Illinois, wild parsnip has become a serious problem in some mesic prairies, and it has been recorded from every county.

**HABITAT**: Although this Eurasian native thrives when growing in rich, alkaline, moist soils, it can survive under almost any condi-tions. Wild parsnip commonly can be found along roadsides, in pastures, and in fields.

**LIFE HISTORY:** Wild parsnip is a perennial that exists as a basal rosette for at least one year and then flowers and dies. Like its relative the carrot, wild parsnip produces a rosette of large, grooved, upright leaves and stores reserves in a large, fleshy taproot during the first year. A hollow flowering stem whose leaves are much smaller is sent up from the center of the rosette in a subse-quent growth season. Wild parsnip

often flowers and sets seed during its second year, although it may not flower until subsequent years. The edible roots of wild parsnip were consumed in ancient Greece and Rome and cultivars are still grown for food today. The root develops its sweet taste after being exposed to cold. Some people are sensitive to the touch of the leaves and soon develop a rash if their skin contacts the leaves or plant sap in the presence of sunlight. A very painful rash can develop that in some people leaves scars that can persisit for several months or longer. Wild parsnip is most irritating at the time of flowering.

**EFFECTS UPON NATURAL AREAS:** Well-established prairies are not likely to be invaded by parsnip, but it can become quite abundant on prairie edges and in disturbed patches within otherwise high-quality prairies. Once established at the edges, parsnip can spread into adjacent high-quality areas.



Photo: © John M. Randall/The Nature Conservancy

**AUTHORS:** Jil Kennay and George Fell, Natural Lands Institute, Rockford, Illinois.

**For more information on wild parsnip, please contact:**
Illinois Nature Preserves Commission, Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/VMG.html
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Wisconsin Department of Natural Resources, Invasive Plant Fact Sheets, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/parsnip.htm

**REFERENCES:**
Eckardt, N. 1987. Element stewardship abstract for Pastinaca sativa - wild parsnip. The Nature Conservancy, Arlington, Virginia. 4 pp.
Fernald, M. L. 1950. Gray's Manual of Botany, eighth edition. American Book Co., New York. 1632 pp.
Jaques, H. E. and R. E. Wilkinson. 1979. How to know the weeds, third edition. William C. Brown Co. Publishers, Dubuque, Iowa. 323 pp.
Jeffery, L. S. and H. J. Lorenzi. 1987. Weeds of the United States and their control. Van Nostrand, Reinhold Co., New York. 257 pp.
Kline, V. M. 1981. Mowing to Control Wild Parsnip (Wisconsin). Restoration and Management Notes 1(1):33.
Kline, Dr. V. M. 1976. Effects of mowing on wild parsnip: six year study (Wisconsin). Restoration and Management Notes 4(2):113.
Mohlenbrock, R. H. 1986. Guide to the vascular flora of Illinois. Southern Illinois University Press, Carbondale. 507 pp.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Wild Parsnip
# *Pastinaca sativa* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - http://www.inhs.uiuc.edu/chf/outreach/VMG/parsnip.html - Vol.1, No. 26 - Authors: Jill Kennay and George Fell - February 6, 1990.**

**Warning**-- Care should be taken to avoid skin contact with the toxic sap of the plant tissues by wearing gloves, sleeves, and long pants. Although eradication of this exotic is desirable from a human safety as well as ecological standpoint, in some situations the best control measure is to do nothing. In high-quality prairies, aggressive growth by other species sometimes can outcompete and eventually displace the parsnip.

## MANUAL AND MECHANICAL CONTROL:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - http://www.inhs.uiuc.edu/chf/outreach/VMG/parsnip.html - Vol.1, No. 26 - Authors: Jill Kennay and George Fell - February 6, 1990.**

The best control is achieved mainly through hand-pulling. Plants should be pulled and removed so that seeds do not develop and plants do not resprout. Wild parsnip is easiest to pull right after a good rain or during a drought when the root shrinks. Another effective practice involves cutting the plant below the root crown before seed set during spring of the second year. The best time is as soon as blooms show, but have not matured. Since the plants do not all flower at once, the area should be rechecked several weeks after the first cutting and the following 2-3 years for newly flowering plants. After a spring burn, wild parsnip rosettes are among the first plants to emerge and may be detected easily and dug out to control its abundance along prairie edges. Seeds do not remain viable if dormant in the ground more than 4 years, so the species can be controlled if there is no outside seed source. Although the practices of hand-pulling, cutting, and digging have been successful in small areas with scattered plants, these practices can become difficult and time-consuming if patches containing hundreds of plants have been allowed to spread unchecked.

Mowing or cutting the base of the stem with a scythe can be effective if it takes place after flowering the second year when the plant is mature and blooming, but before seed set. Parsnip must be remowed or recut often and checked later for small bloom shoots near the ground. Poorly timed mowing, as is likely along roadsides, may increase both number of seedlings and percentage surviving to maturity. Mowing probably favors parsnip maturation by allowing more sunlight to reach immature parsnip plants, which are too low to be damaged by the mower. Mowing also reduces the density, height, and flowering of other species that are potentially good competitors against parsnip, such as common goldenrod.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Wild Parsnip - http://tncweeds.ucdavis.edu/esadocs/pastsati.html - Author: Nancy Eckardt - July 23, 1987.**

Wild parsnip can become abundant along roadsides that are regularly mowed as mowing seems to encourage the production of flowering plants. If mowing occurs too early (in June or early July), the plants may resprout and still have time to flower and set seed; if too late in July, the primary umbel may have mature seeds that will germinate after cutting. Mowing also stresses other species that have the potential to be good competitors against parsnip, such as Solidago spp. Kline (1986) tested annual mowing of parsnip in July before seed set over a six- year period and observed increases in the abundance of flowering plants in the mowed plots, but a steady decline in parsnip density in the unmowed control plot. The common goldenrod, Solidago altissima, was abundant in all plots at the start of the experiment. The July mowing reduced density,

height, and flowering of the goldenrod, allowing more sunlight to reach immature parsnip seedlings. The steady decline in parsnip density in the unmowed plot suggested that in situations where other plants are able to offer competition, the best parsnip control measure is to do nothing (Kline 1986).

Where P. sativa occurs on a recovering prairie, the best treatment may be to simply encourage good prairie growth. For example, prescribed burning encourages the growth of native grasses, which in turn outcompete and eventually displace the wild parsnip (Kline 1987).

For small patches, weeding with a shovel is the best control measure. Flowering plants should be chopped off just below ground level before seed set. Care should be taken to avoid contact with the plant tissues. Wear gloves, long pants, and sleeves. Since the plants do not all flower at once, the area should be rechecked several weeks after the first cutting. The vegetative rosettes can also be dug up if enough labor is available, otherwise, the area should be revisited the following year to remove any newly flowering plants.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - Wild Parsnip - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/parsnip.htm**

The best way to control wild parsnip is early detection and eradication. A very effective control method is to cut the entire root just below ground level with a sharp shovel or spade. Cutting below ground level prevents resprouting. In some soil types in wet conditions, the plants can be pulled out of the ground by hand. All seeds must be removed from the site and disposed of in a landfill or by burning.

If the population is too large to hand-cut or pull, a power brush-cutter can be used just after peak flowering and before the seeds set. Plants may resprout when cut above the ground, and should be cut again a few weeks later to prevent flowering. Cutting done after seed set will greatly reduce the likelihood that the plants will be able to resprout and flower. Plants cut at this time must all be gathered and removed from the site to prevent mature seed from developing and falling to the ground. Another effective way to eliminate reseeding is to hand-collect all seeds after they have set. If control of flowering or seeding plants is carried out over several years, the population will decrease as the seed bank is depleted.

## PRESCRIBED FIRE:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - http://www.inhs.uiuc.edu/chf/outreach/VMG/parsnip.html - Vol.1, No. 26 - Authors: Jill Kennay and George Fell - February 6, 1990.**

Burning does not successfully control parsnip be-cause it removes litter and taller plants, providing favor-able condi-tions for parsnip rosettes to develop. However, periodic burning maintains the vigor of native plants, allowing them to compete with parsnip.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Wild Parsnip - http://tncweeds.ucdavis.edu/esadocs/pastsati.html - Author: Nancy Eckardt - July 23, 1987.**

Burning removes litter and taller plants allowing parsnip rosettes to develop rapidly. When present, wild parsnip rosettes are among the first plants to green up after an early spring burn and they become easy to detect and dig up with a shovel.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - Wild Parsnip - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/parsnip.htm**

Burning does not seem to impact the plants themselves -- they quickly resprout. However, in the darkened soil following a burn, these rosettes are easy to recognize and can be controlled by hand-digging. Prescribed burning stimulates increased growth in prairie species that may potentially decrease parsnip populations through competition.

## BIOCONTROL:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - http://www.inhs.uiuc.edu/chf/outreach/VMG/parsnip.html - Vol.1, No. 26 - Authors: Jill Kennay and George Fell - February 6, 1990.**

The parsnip webworm damages some individual plants severely, but is not known to eradicate whole patches and is not likely to be useful as a biocontrol agent.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Wild Parsnip - http://tncweeds.ucdavis.edu/esadocs/pastsati.html - Author: Nancy Eckardt - July 23, 1987.**

The parsnip webworm damages some individual plants severely, but is not known to devastate whole patches and is not likely to be useful as a biocontrol agent (Martin 1987).

## CHEMICAL CONTROL*:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - http://www.inhs.uiuc.edu/chf/outreach/VMG/parsnip.html - Vol.1, No. 26 - Authors: Jill Kennay and George Fell - February 6, 1990.**

If mechanical methods have failed to control wild parsnip or are not feasible, a 2% spot application of the herbicide Roundup (glyphosate) to basal rosettes is a recommended treatment. Roundup should be applied to individual plants with a hand sprayer in late fall after most native vegetation is dormant. Late fall application minimizes the potential harm to nontarget species. It may be necessary to treat the same area again annually until missed plants and plants originating from the seed bank are eliminated. Roundup is a nonselective herbicide (kills all vegetation)

and should not be used in high-quality natural communities during the growing season because of the possibility of harming nontarget plants.

The herbicide 2,4-D (available under a variety of trade names) mixed according to label directions and applied to individual parsnip basal rosettes between March-May or August- October is effective. This herbicide should only be used on buffer or severely disturbed sites, and not in high-quality natural communities if it is applied during the growing season. Repeated early spring applications of this chemical before the flower stalk begins to elongate will reduce infestation of wild parsnip.

Care should be used to avoid contacting nontarget plants when applying either herbicide. **Do not spray so heavily that herbicide drips off the target species**. Native nontarget species will be important in recolonizing the site once the parsnip dies. The herbicide should be applied while backing away from the treated area to avoid contact with wet herbicide. By law, herbicides only may be applied according to label instructions and by licensed herbicide applicators or operators when working on public properties.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - Wild Parsnip - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/parsnip.htm**

Chemical controls are effective, but should be used sparingly on quality natural areas. The best method is to burn the site, then follow with spot application of 1-3% active ingredient glyphosate. Immediately after a burn, wild parsnip is one of the first plants to green. Glyphosate can be spot applied to the basal rosette of the parsnip with little effect on dormant species.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Beefsteak plant
## Perilla frutescens L. (Britt.)

**SOURCE:** The following description was taken from Rhoads and Block, The Plants of Pennsylvania: An Illustrated Manual, 2000. Images were taken from www.invasive.org and plants.usda.gov.

**DESCRIPTION**: Beefsteak plant is an herbaceous annual that invades a variety of habitats. It has been introduced as a cultivated plant and used medicinally. The plant can reach heights of 10-40 cm. Leaves are egg-shaped, 8-15 cm and generally purple underneath. Flowers are small, purplish to white and paired along the spike-like inflorescence.




Photo: Jil M. Swearingen, USDI National Park Service, www.forestryimages.org.



Photo: Douglas Ladd @ USDA-NRCS PLANTS Database / USDA SCS. 1989. Midwest wetland flora: Field office illustrated guide to plant species. Midwest National Technical Center, Lincoln, NE.

**For more information on beefsteak plant, please contact:**
Mid-Atlantic Exotic Pest Plant Council, http://www.ma-eppc.org/
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm.
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA-NRCS PLANTS Database, http://plants.usda.gov/.

**REFERENCES:**
Rhoads A.F. and T.A. Block. 2000. The Plants of Pennsylvania: An Illustrated Manual. University of Pennsylvania Press. 1061 pp. USDA-NRCS PLANTS Database, http://plants.usda.gov/.

**Management and Control Information**

Back to List




## Species Management and Control Information

### Beefsteak plant
### *Perilla frutescens* L.

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

**Management information for this species was unavailable at time of tutorial development.**

Return to List




## Reed canary-grass
## *Phalaris arundinacea* L.

**SOURCE**: The following information on this species is taken from the Delaware River Invasive Plant Partnership, Invasive Plant Fact Sheets at http://www.paflora.org/DRIPP.html. Modifications include additional pictures from www.invasives.org and http://tncweeds.ucdavis.edu/index.html. Check the website links for future updates. The last update for this fact sheet is April, 2002.

**DESCRIPTION**: Reed canary-grass is a cool-season grass that grows 3-8 feet tall. It forms dense, monospecific stands in wet meadows, riparian areas, and marshes; spreading by means of stout rhizomes. Established plants of reed canary-grass can tolerate prolonged periods of inundation. Other species that might be confused with reed canary-grass include: common reed (Phragmites australis), Canada bluejoint (Calamagrostis canadensis), or orchard grass (Dactylis glomerata). The long, membranous ligule of reed canary-grass can be helpful in distinguishing it from the others. In addition, unlike common reed, the stems of reed canary-grass do not remain standing through the winter.
Height - Flowering stems grow from 3-7 feet tall.
**Stem** - The stem is smooth, green, and erect.
**Leaves** - Leaves are 4-8 inches long and about ½ inch wide; the ligule is unusually large - up to ½ inch long.
**Flowers** - Flowering occurs in June and July.
**Fruit and seed** - The 3-10 inch long branched inflorescence is at the top of an erect stem. It is green with a purplish tinge; although narrow at first, it opens up when in flower. The plume contracts and becomes light tan in color in the fruiting stage. Seeds germinate most readily immediately after they mature.
**Roots** - Reed canary-grass spreads by rhizomes to form large clonal colonies.




Photo: Chris Evans, Univ. of Georgia, www.forestryimages.org

Photo: © John M. Randall/ The Nature Conservancy

Photo: © Barry M. Rice/ The Nature Conservancy

**DISTRIBUTION AND HABITAT:** Reed canary-grass is a circumboreal species; in North American it grows from Newfoundland to Alaska and south to North Carolina, Kansas, and California. Why it has become so invasive is not fully known, but may be related to the development of agronomic cultivars with increased vigor and drought tolerance. As early as the 1830s efforts were underway in New England to develop superior forms of reed canary-grass for use as a forage crop. Whether we are also dealing with several naturally occurring genotypes, as we have learned recently is the case with common reed (*Phragmites australis*), has not been determined. Reed canary-grass occurs throughout Pennsylvania in marshes, alluvial meadows, shores, and ditches. It dominates the extensive riparian flats along the upper Delaware River. EFFECTS OF INVASION: Reed canary-grass forms dense, monospecific stands in open wetlands, wet meadows, riparian areas, and shores. It effectively excludes all other plant species, causing greatly decreased biological diversity in wetland communities.

Photo: © John M. Randall/ The Nature Conservancy



**REPRODUCTION AND METHODS OF DISPERSAL:** Both seed production and vegetative spread by means of rhizomes contribute to the success of reed canary-grass. Seeds are wind-disseminated; both seeds and rhizome fragments are undoubtedly also transported by water. Reed canary-grass produces few viable seeds unless cross pollination between clones occurs.

**NATIVE ALTERNATIVES FOR LANDSCAPE USE:** Reed canary-grass should not be planted in wetland restoration programs, native grasses and other monocots that do not exhibit invasive tendencies include: big bluestem (Andropogon gerardii), meadow foxtail (Alopecurus pratensis), Canada wild rye (Elymus canadensis), hairy wild rye (Elymus villosus), bottlebrush grass (Elymus hystrix), fowl mannagrass (Glyceria striata), rattlesnake grass (Glyceria canadensis), slender mannagrass (Glyceria melicaria), Indian grass (Sorghastrum nutans), soft rush (Juncus effusus), soft-stem bulrush (Schoenoplectus tabernaemontana), wool-grass (Scirpus cyperinus), and many sedges (Carex spp.).

**AUTHORS:** Ann F. Rhoads and Timothy A. Block, Morris Arboretum, University of Pennsylvania.

**For more information on reed canary-grass, please contact:**
Delaware River Invasive Plant Partnership, http://www.paflora.org/DRIPP.html
Illinois Nature Preserves Commission, Vegetation Management Guidelines, http://www.inhs.uiuc.edu/chf/outreach/VMG/rcanarygr.html
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Ohio Department of Natural Resources, Division of Natural Areas and Preserves, Invasive Plant Fact Sheet, http://www.dnr.state.oh.us/dnap/invasive/6canarygrass.htm
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Wisconsin Department of Natural Resources, Invasive Plant Fact Sheets, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/reed.htm

**REFERENCES:**
Apfelbaum, Stephen I. and Charles E. Sams. 1987. Ecology and control of reed canary-grass (Phalaris arundinacea L.). Natural Areas Journal 7 (2): 69-74.
Merigliano, M. F. and P. Lesica. 1998. The native status of reed canary-grass (Phalaris arundinacea L. ) in the inland Northwest, U.S.A. Natural Areas Journal 18: 223-230.
Rhoads, Ann Fowler and Timothy A. Block. 2000. The Plants of Pennsylvania: An Illustrated Manual. University of Pennsylvania Press, Philadelphia, PA.
Rhoads, Ann Fowler and William McKinley Klein. 1993. The Vascular Flora of Pennsylvania: Annotated Checklist and Atlas. American Philosophical Society, Philadelphia, PA.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Reed canary-grass
# *Phalaris arundinacea* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheet - Reed canary-grass - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Whatever means is used to eliminate reed canary-grass, it is essential to replant immediately with something that can out compete any residual plants or seeds, and retard erosion.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract) - Phalaris arundinacea - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Kelly E. Lyons - December 22, 1998.**

As with most invasive species the best management strategy is prevention (Lesica, 1997). Control is difficult due to the rhizomatous nature of the species and may require herbicide treatment for several years. In addition, few herbicides may be used in wetlands or areas near running water, where reed canarygrass is usually most troublesome. When reed canarygrass is eliminated, there may be a danger of soil erosion if other species fail to cover the area quickly. Reed canarygrass is itself especially good at invading denuded areas and one way to prevent it from becoming established is to control erosion and replant eroded sites with competitive native species (Hoffman & Kearns, 1997).

Hutchison (1992) suggests that reed canarygrass should never be completely removed from a site because it is impossible to tell the difference between native and nonnative strains. If you determine that the plants are rapidly spreading, extremely competitive, and displacing many native species at a rapid rate, however, consider complete eradication as a goal.

A mixed management strategy may prove most successful for control of reed canarygrass. For example, Paveglio & Kilbride (1996) compared five different treatments: disking, mowing, early and late treatments with glyphosate herbicide, late glyphosate treatment alone, and early glyphosate treatment + disking. Glyphosate was applied as a 0.5% solution of Rodeo at 5.26L/ha (2.25 quarts/A) plus LI-700 surfactant. The early glyphosate treatment plus disking suppressed reed canarygrass and promoted the growth of other species. The early and late glyphosate application was also effective, providing 99% control in the first year.

The type of control chosen should depend on site characteristics. Where reed canarygrass is mixed with other species, burns or mechanical removal will be more effective as the area will be readily occupied by species that could potentially outcompete reed canarygrass and control erosion. Non-selective herbicides like glyphosate are most effective on sites that are totally dominated by reed canarygrass (Henderson 1987). Any patch left denuded is subject to erosion and re-invasion.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/reed.htm**

Reed canary grass is difficult to eradicate; no single control method is universally applicable. In natural communities, mechanical control practices are recommended. In buffer areas and in severely disturbed sites, chemical and mechanical controls may be used. If herbicide is used, care should be taken to prevent contact with non-target species. Any control technique to reduce or eliminate reed canary grass should be followed by

planting native species adapted to the site. As reed canary grass can enter a wetland area from eroding hillslopes, erosion control and catch-basins around a preserved wetlands are appropriate preventative measures.

## MANUAL AND MECHANICAL CONTROL:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheet - Reed canary-grass - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Hand pulling is practical in small areas that are newly infested. Follow-up will be needed over 4-5 years, as fragments of rhizome will inevitably remain and grow. Mowing 5 times in a season has been found to be effective. Discing or plowing is another way to control reed canary-grass; however, many places where it is a problem are too wet for this to be practical. Flooding will kill the seeds, which cannot survive prolonged inundation, but established rhizomes are unaffected.

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Reed Canary Grass - http://www.inhs.uiuc.edu/chf/outreach/VMG/rcanarygr.html - Vol.1, No. 19 - Author: Max Hutchison - February 6, 1990.**

Hand removal for control may be feasible in small stands. There is evidence that hand chopping the culms at flowering time may kill small clones.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plant Fact Sheet # 6 - Reed canary-grass - http://www.dnr.state.oh.us/dnap/invasive/6canarygrass.htm - January 8, 2003**

In smaller patches, hand-pulling or digging may be effective. Mowing can be used to control seed production by mowing in early to mid-June and early October before seed matures. This also exposes the soil to light which will promote the growth of other species. Discing or plowing can also be used to control a well-established population.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract) - Phalaris arundinacea - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Kelly E. Lyons - December 22, 1998.**

MECHANICAL: Removal of reed canarygrass by hand-pulling is practical only for small stands and requires a large time commitment (Hutchison, 1992). Henderson (1987) found hand-pulling was effective if done over the entire population 2-3 times per year for five years.

Hoffman and Kearns (1997) suggest covering reed canarygrass infestations with black plastic. They claim that for this method is to be successful, light levels should be reduced to less than 40% of normal intensity and the plants should not be allowed to grow beyond the plastic. (Shoots emerging beyond the edges of the covering will provide food to covered rhizomes.) Following successful control with black plastic, the area can be seeded with local, native species. However, this method was found to have little success by Apfelbaum & Sams (1987), who observed plants persisting even after two years under cover.

COMPETITIVE CROPS/RESTORATION: Native grasses and forbs are the best plants to use as competitors. Seeds can be collected and raked into the soil after reed canarygrass control. Competitive crop management has been most effective when used in conjunction with prescribed burning (Hutchison, 1992).

Desert saltgrass (Distichlis stricta) may be used as a "replacement species" in alkaline or saline soils where erosion may pose a problem after herbicide application and subsequent removal of reed canarygrass (Marquis et al., 1984). While native to the United States, desert saltgrass is found primarily in salt marshes and alkaline habitats (Hickman 1993) and may grow poorly outside of these habitats.

CUTTING: Cutting can effectively control reed canarygrass. Reed canarygrass was practically non-existent in fields after they had been cut five times in one season, even though the fields that had started out as roughly equal mixtures of reed canarygrass, Kentucky bluegrass (Poa pratensis), timothy (Phleum sp.) and mixed clovers (Trifolium spp.) (Wilkins and Hughs, 1932). If the plots were cut only twice during the season, reed canarygrass persisted.

DISCING/PLOWING: Reed canarygrass is quite sensitive to discing or plowing, and in some situations other plant species readily outcompete reed canarygrass afterwards. Paveglio and Kilbride (1996) found that an early season herbicide application (Rodeo at 5.26liters/ha) followed by discing three times late in the season suppressed reed canarygrass. The herbicide application prior to discing causes reed canarygrass rhizomes to deteriorate. Applying an herbicide prior to discing eases the task of discing and suppresses post-discing seedling emergence. Later applications of herbicide may be necessary to control seedlings.

ALTERATION OF WATER LEVELS: When wetlands have had their water levels lowered (for example, by ditching) restoration of original water levels may control reed canarygrass. Vegetative portions of reed canarygrass are generally regarded as intolerant of prolonged inundation (Hutchison, 1992) but some studies have found opposing results where rhizomes tolerate prolonged inundation (LeFor, 1987).

The seeds of reed canarygrass are generally short-lived when inundated. Long periods of inundation may serve to eliminate reed canarygrass seeds from soils. Comes et al. (1978) found most reed canarygrass seeds decomposed and/or germinated after only three months but germination rates of some seed remained high for 3-12 months after inundation. After 48 months of inundation, however, seeds of reed canarygrass no longer germinated.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/reed.htm**

Small, discrete patches may be covered by black plastic for at least one growing season; the bare spot can then be reseeded with native species. This method is not always effective and must be monitored because rhizomes can spread beyond the edge of the plastic.

Mowing twice yearly (early to mid-June and again in early October) may help control reed canary grass by removing seed heads before the seed matures and exposing the ground to light, which promotes the growth of native wetland species. Discing the soil in combination with a mowing or burning regime may help by opening the soil to other species. Hand-pulling or digging may work on small stands in the early stages of invasion. Grazing can enhance diversity, although it will not control reed canary grass. A bulldozer can be used to remove reed canary grass and rhizomes (12-18" deep), after which native species should be seeded. Discing or plowing can also be employed in this manner.

In small areas with few natives, another method involves repeated cultivation for one full growing season followed by dormant seeding near the first-frost date. Disrupting the plant roots every two to three weeks weakens the remaining plants and depletes the seed bank. When combined with spot herbicide application in sections too wet for early or late cultivation, results after two years have been good. Frequent and continued cultivation is important since one or two cultivations would simply cut the roots up and increase the number of individual plants.

## PRESCRIBED FIRE:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheet - Reed canary-grass - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Burning also works, however, timing is important; late spring or late fall are recommended.

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Reed Canary Grass - http://www.inhs.uiuc.edu/chf/outreach/VMG/rcanarygr.html - Vol.1, No. 19 - Author: Max Hutchison - February 6, 1990.**

RECOMMENDED PRACTICES IN NATURAL COMMUNITIES OF HIGH QUALITY: Fire can help control the spread of reed canary grass and keep it out of high quality wetlands. Repeated late autumn or late spring burning for several years can control this species. Annual burning may be needed for 5-6 years before good control is apparent. Burning is most effective where other species are present or in the seed bank, since fire allows native, fire-adapted species to compete successfully. Fire is not very effective in dense monocultures of this grass where seeds or plants of native species are absent.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plant Fact Sheet # 6 - Reed canary-grass - http://www.dnr.state.oh.us/dnap/invasive/6canarygrass.htm - January 8, 2003**

Although prescribed burning can be effective, it must be repeated annually for 5 or 6 years. Timing may be difficult due to fluctuating water levels and the growth stage of the plants at burn time. A combination of these measures used together may improve results.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract) - Phalaris arundinacea - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Kelly E. Lyons - December 22, 1998.**

Fire is an effective form of control for reed canarygrass in highly productive wetlands. Some wetland species are unable to out-compete reed canarygrass without prescribed burning (Hutchison, 1992). Fire should be reserved for sites containing a healthy seed bank of fire-adapted native species that will readily colonize the area after a burn.

Apfelbaum & Sams (1987) describe a study that successfully contained reed canarygrass in low quality areas of a wetland using a two-year or three-year burn rotation cycle. Prescribed fire may be required for five or six years. The timing of burns is important. Henderson (1991) found that early spring burns accelerate the spread of reed canarygrass while late spring burns weakened reed canarygrass stands. Unfortunately, these late-season burns can harm other species. Late autumn burns are also beneficial.

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/reed.htm**

Prescribed burns in late spring or late fall may help reduce the reed canary grass population if repeated annually for 5 to 6 years. However, these fires are difficult to conduct due to water levels and/or the greenness of the grass at the time of burning. The application of 1.5% active ingredient solution of glyphosate will "brown off" reed canary grass enough to conduct prescribed burns. Burning is also ineffective in dense stands of reed canary grass that lack competition from native, fire-adapted species in the seed bank. A late-spring burn followed by mowing or wick-applying glyphosate to the emerging flowering shoots will eliminate reed canary grass seed production for that year.

## GRAZING:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract) - Phalaris arundinacea - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Kelly E. Lyons - December 22, 1998.**

Grazing may be effective means for controlling reed canarygrass but the palatability of reed canarygrass is questionable--the genus Phalaris is notoriously unpalatable and an illness associated with the affects of consumption is called 'Phalaris staggers' (Marten et al., 1976). Grazing can also be inappropriate in wetland settings (Hutchison, 1992). However, Haslam (1973) suggests employing wetland meadows for the production of "marsh hay." Ponies are preferable grazers as they are more agile in waterlogged soils and may be left unattended while cattle and sheep may get stuck in the mud and perish.

Palatability decreases with plant maturity (Marten et al., 1976; Gomm, 1979) and continuous grazing may decrease palatability. Plants' chemical and physical defenses may be induced by herbivory, but cutting often fails to elicit the same defensive response (Agrawal, 1998). The literature does not address the response of reed canarygrass alkaloid concentrations in response to grazing.

## BIOCONTROL:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheet - Reed canary-grass - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

No biological control options are currently known.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plant Fact Sheet # 6 - Reed canary-grass - http://www.dnr.state.oh.us/dnap/invasive/6canarygrass.htm - January 8, 2003**

There are currently no biological control methods in use for reed canary grass.

## CHEMICAL CONTROL*:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheet - Reed canary-grass - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Glyphosate will control reed canary-grass; but, because it often grows in or near water, only formulations approved for aquatic use (Rodeo) should be utilized.

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Reed canary-grass - http://www.inhs.uiuc.edu/chf/outreach/VMG/rcanarygr.html - Vol.1, No. 19 - Author: Max Hutchison - February 6, 1990.**

RECOMMENDED PRACTICES IN BUFFER AND SEVERELY DISTURBED SITES: Certain herbicides are effective where there is no real concern for damage to surrounding native species. Rodeo, a formulation of glyphosate designed for use in wetlands, will kill reed canary grass, especially young plants, when applied to foliage according to label recommendations. Rodeo should be applied in early spring when reed canary grass is green and most native wetland species are still dormant. The area should be checked after spraying, and any surviving reed canary grass should be sprayed the following spring. Dalapon and Amitrol also reportedly kill canary grass, although no Illinois natural area managers were found that have experience with these herbicides. All 3 herbicides are licensed for use in aquatic areas. **Rodeo and Amitrol are nonselective herbicides that will kill all vegetation contacted**. Dalapon selectively kills grasses and monocots, but not broadleaf plants. Spraying foliage with Roundup (a formulation of glyphosate) mixed according to label instructions and subsequent burning of dead residue has been moderately effective in northern Illinois. However, Roundup is not licensed for use in aquatic areas and should only be used in areas without standing water. When using any herbicide, precautions should be taken to avoid contacting nontarget species. **Do not spray so heavily that herbicide drips off the target species**. The herbicide should be applied while backing away from the treated area to avoid contacting wet herbicide. By law, herbicides only may be applied as per label instructions and by licensed herbicide applicators or operators when working on public properties. Where practical, it can be useful to sow in seed of nearby native grasses and forbs after reed canary grass has died (due to control efforts) or gone dormant. Seed of nearby native grasses and forbs should be collected when ripe and then raked into the sod as soon as the reed canary grass has died.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plant Fact Sheet #6 - Reed canary-grass - http://www.dnr.state.oh.us/dnap/invasive/6canarygrass.htm - January 8, 2003**

Herbicides, such as Accord® or Glypro®, can be applied to control reed canary grass. Fusilade DX®, a grass specific herbicide, can be applied in non-wetland areas. Herbicide should be applied in early spring when nontarget species are still dormant. Removal of the previous year's growth to expose the new green shoots aids effectiveness of the chemical and minimizes the amount needed. Foliar application of Glypro® to larger monocultures of reed canary grass can be effective. Chemical treatments following mowing in the fall season can help control this grass as well.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract) - Phalaris arundinacea - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Kelly E. Lyons - December 22, 1998.**

Several herbicides have been tested and proven effective for controlling reed canarygrass. The most effective include glyphosate and fluazifop butyl. When applying herbicides, take precautions to avoid contact with non-target species.

In general, reed canarygrass is more effectively controlled by herbicides on upland, drier areas of a site (Hodgson, 1968). In one experiment, upper areas on a canal ditch had 100% control while in the lower, wetter areas only 50% of the reed canarygrass was removed. Bruns (1973) was also unable to kill all shoots at the waterline. This was due to dilution of the herbicide when applied at or near the water and removal of the herbicide from the soil by water.

Reed canarygrass is rarely fully eradicated and yearly, or even monthly, attention to the control of this weed is necessary. Hodgson (1968) found consecutive, yearly treatments were required to control reed canarygrass. Hoffman & Kearns (1997) suggest that old vegetation be removed before herbicides are applied to ensure that the herbicide contacts new growth. Where large populations of reed canarygrass are eliminated, seeds of other species should be sown to increase competition with resprouts and prevent erosion.

**FLUAZIFOP-D (FUSILADE, HORIZON)**
Apply fluazifop at 0.28-0.42 kg ai/ha (0.25-0.375lb ai/A) (1 to 1.5 pint product/A) to actively growing reed canarygrass with 1% v/v crop oil concentrate or 0.25% v/v nonionic surfactant. Fluazifop acts very slowly (2-4 weeks before results are seen). Do not apply to stressed grasses. If regrowth occurs, repeat application. Do not use crop oil concentrate if treating reed canarygrass ornamentals. Do not apply if rainfall is expected within 1 hour (William et al., 1997).

**GLYPHOSATE (RODEO OR ROUNDUP)**
Rodeo is a formulation of glyphosate that is specifically designed for use in aquatic habitats. It will kill reed canarygrass if applied to young plants according to the manufacturer's directions. It should be applied in early spring when reed canarygrass is just sprouting and before other wetland species germinate. Rodeo is nonselective and should be applied only in areas where damage to other species is unlikely (Hutchison, 1992). Roundup is not registered for use in aquatic areas and is designed for use only where there is no standing water. Rodeo and Roundup (1.4 to 2.8 kg ai/ha; 1.2 to 2.25 lb ai/A) should be applied to actively growing plants at early heading or in the fall. Follow the manufacturer's instructions regarding surfactants (William et al., 1997).
An application of Roundup according to the manufacturer's recommendations and combined with the burning of dead residue was moderately effective in Illinois (Hutchison, 1992).
In Prosser, Washington, glyphosate was applied to seedlings of three species (reed canarygrass, red top and creeping fescue) at three, five, seven, and ten weeks post-emergence and to 15 month old plants in the spring. Applied at 1.1 kg/ha, a decrease in reed canarygrass at all age stages was observed without causing much damage to red top and fescue. For 15 month old plants, glyphosate applied at 2.2 kg/ha yielded up to 95% control (Comes, 1976). · At TNC's Middle Fork John Day preserves, Roundup was reported to be an effective control measure at high concentrations (Youtie, personal communication).

**SULFOMETURON (OUST)**
Apply at 26-43 g ai/ha (3 to 5 oz product/A) to preemergent or early postemergent plants. Oust is labelled for use on non-cropland only. Do not apply to frozen ground. Maintain constant agitation while mixing in spray solutions. Add 0.25% v/v nonionic surfactant to increase the activity of postemergence applications. Powder, dry soils and light, sandy soils should not be treated when there is little likelihood of rainfall after treatment (William et al., 1997).

**Wisconsin Department of Natural Resources - Invasive Plant Fact Sheets -**
**http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/reed.htm**

Small, scattered clones (2 feet in diameter) can be controlled by tying the stems together just before flowering, cutting them, and applying glyphosate in a 33% active ingredient (a.i.) solution to cut stems.

A formulation of glyphosate designed for use in wetlands will kill reed canary grass (especially young plants) when applied to foliage. Apply in early spring when most native plant species are dormant. Any herbicide application should be done only after removing dead leaves from the previous year in order to maximize growing shoot exposure and to minimize herbicide use. A 5% a.i. solution of glyphosate formulated for use over water applied as a foliar spray will kill reed canary grass. Two herbicidal applications may be necessary to ensure complete coverage. Herbicide applied with a wick applicator attached to a tractor affects taller stands of reed canary grass without impacting the shorter vegetation.

A late mowing in mid-September, followed by the application of 5% glyphosate a.i. in October (after big bluestem is dormant) can help to control reed canary grass.

Because reed canary grass productivity is reduced by shade, planting native shrubs or wetland trees in areas of chemically-treated grass may be effective.

While herbicide kills reed canary grass, the seed bank may germinate and recolonize the site. Several herbicidal application may be necessary to inhibit seed bank recolonization. After the first application of herbicide has killed living plants, disturbance of the soil can encourage seed bank germination. When this occurs, the site can again be treated with herbicide to deplete the seed bank.

An alternative method involves wick application of glyphosate in the first to third weeks of June, followed by a late June to mid-July burn. This technique reduces reed canary grass cover, depletes the seed bank, and stimulates native seed banks.

In non-aquatic environments, DalponTM and Trichloracetic (TCA)TM effectively treat reed canary grass when applied in late fall or early winter. Both are soil sterilizing herbicides that must be sprayed on dried foliage in a rate of 20 to 40 lbs. per acre. Aquatic systems may also be treated with DalponTM for control up to two years. DalponTM is weakly cationic and is not absorbed by substrates the way most herbicides are.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

## OTHER CONSIDERATIONS:

**Illinois Nature Preserves Commission - Vegetation Management Guidelines - Reed Canary-grass -**
**http://www.inhs.uiuc.edu/chf/outreach/VMG/rcanarygr.html - Vol.1, No. 19 - Author: Max Hutchison - February 6, 1990.**

FAILED OR INEFFECTIVE PRACTICES:

- · hand control: very slow and too labor-intensive for large stands.
- mowing: probably not effective as a control measure.
- herbicides: probably none that are selective enough to be useful in high quality areas.
- grazing: probably not a practical method of control in wetland areas where canary grass usually is a problem.
- tillage: not usually practical in wetlands and not appropriate for high quality sites.
- restoring water levels: many Illinois wetlands are drier now than historically, and restoring water levels needs more research.
- biological controls: none known that are feasible in natural areas.
- introduction of competitive species: probably few native species can compete with reed canary grass in wetlands if burning is not used also. It reportedly will even crowd out cattails. Prescribed burning allows native species that are present or seeded-in to compete successfully.

- heavy equipment: removal with construction equipment is ineffective, as reed canary grass responds quickly by growing back from rhizomes and seeds remaining in the soil. Use of heavy equipment is not appropriate in good quality natural communities.

**The Nature Conservancy - Invasive Species Initiative - Reed canarygrass: Control and Management in the Pacific Northwest - http://tncweeds.ucdavis.edu/esadocs/phalarun.html - Author: Mandy Tu - June 7, 2004.**

Control Summary
There is no immediate one-year "fix" to convert a RCG infestation into a native community, but much can be accomplished within 2 to 3 years. Continued monitoring and follow-up treatments will be required for up to 5 to 10 years to prevent reinvasion. Reed canarygrass is difficult to control due to its persistent rhizome system and its ability to reproduce both vegetatively and sexually. However, depending on available time and resources, even highly infested areas can be restored to more desirable vegetation. Be sure to use an adaptive management process to determine your management objectives, to develop your work plan, and to be able to continually update that plan as results of your management actions become apparent. The successful restoration of your wetland system from RCG will likely involve five steps:
1. The control/removal/kill/local eradication of the existing RCG plants and rhizome system.
2. Exhausting the RCG seed bank.
3. Depending on how long the RCG infestation has been in your site, active restoration (replanting or reseeding of desirable vegetation) may be required. If your RCG infestation has not been at that site for over 5 to 10 years, there may be enough of a remnant native seedbank to allow passive restoration.
4. Prevention of new seeds or stem fragments from entering your managed area, and/or changing those conditions that facilitated RCG invasion in the first place.
5. Finally, continued monitoring and follow-up treatments are necessary for lasting results.

There are a variety of methods available for the control of RCG. Which method or combination of methods you choose will depend ultimately on your management goals and objectives (are you trying to fully restore a Puget Sound wetland or just add structural diversity to a Willamette Valley riverbank?). Also, how many resources you are willing to invest and for how long, what resources do you already have available, and the size, distribution, and location of your RCG infestation will all determine which option you should choose to manage RCG. Unless you only have a few small isolated patches, the long-term successful management and control of RCG will require a multi-year commitment.

Prevention
Prevention of new invasions is the most efficient and cost effective method of invasive species management and control, and the prevention of new RCG infestations are no exception to this rule. Maintaining a healthy community of native or otherwise desirable plants, taking care to not disperse RCG seed or propagules, and carefully monitoring your managed area periodically (especially along roadside ditches and other disturbed areas) and eradicating small RCG populations as soon as possible and in neighboring lands can greatly benefit your RCG management efforts. Another prevention method is to work to change those environmental conditions that allowed RCG invasion in the first place. Recent research completed from Wisconsin and Minnesota have shown that when levels of available soil nutrients (namely nitrogen) are reduced via carbon enrichment, a native sedge, Carex hystericina, is able to competitively suppress the growth of RCG. Sustaining a mosaic of microtopographies (by preventing sediment accumulation) facilitates native species richness, and maintaining complex herbaceous canopies also work to prevent RCG infestation, since RCG seed germination is dependent on amounts of light penetration.

Manual & mechanical methods
Digging: Isolated plants or small patches of RCG can successfully be removed by digging out and removing the entire root mass. Removal is easiest when the soil is moist. Be sure to remove all rhizomes and roots, as small rhizome fragments can resprout. Properly dispose of plant material, since rhizomes and stems can develop new roots if inundated, or if kept in contact with moist ground. Be sure to follow-up to catch any resprouted stems.

Mowing/Cutting: Mowing or cutting (using a mower, brush cutter, weed eater, tractor-drawn mower, machete, etc.) by itself will not kill RCG. In fact, if RCG is mowed only once or twice per year, it actually stimulates additional stem production. Continued mowing (5x or more per year) for 5 to 10 years is reported as successful in controlling RCG, but this has not been demonstrated on a large scale in the PNW. Mowing can be used in combination with another control method, such as followed by a herbicide application, for good control. Additionally, mowing prior to or at the onset of flowering can eliminate seed set for that year. So, you can choose to mow RCG for several years to eliminate the seed bank, and then a final mow followed by herbicide application to eliminate mature RCG. Mowing can also facilitate the installation of shade cloth, or be used as a pre-treatment for tillage, since it will remove or break up the thick layer of dead litter.

Tillage/Cultivation: The use of large tillage machinery can successfully eliminate RCG if combined with a proper flooding regime. The USFWS Refuges at Ridgefield (near Vancouver, WA) and Finley (near Corvallis, OR) have successfully used a combination of tillage + flooding to convert large RCG infestations into native wetland habitats. This method, however, requires the use of large, expensive equipment,and requires the ability to manipulate water levels. Additionally, use of tillage to manage RCG assumes that you have no species or communities of concern that you are trying to preserve at the site. If you are working in a sensitive area or in a relatively intact native system, this may not be a viable option. The purchase of the large tillage equipment (48-inch tillage plates and tractor) can be prohibitively expensive, but it may be available locally for rental or borrowing. To eliminate large, dense RCG infestations using tillage + flooding, you should till through the RCG sod layer as soon as it is possible in the field season (usually, as soon as it is dry enough). The initial tillage may require several passes of the equipment, since the RCG sod layer may be thick and tough. Let the exposed stems and rhizomes dry-out. You will need to till several times during the field season to break-up and dry all rhizome fragments (until you have nothing left but broken-up clods of soil). Finally, when the winter flooding begins, close floodgates and keep the entire area inundated at least 18 inches deep through late spring (late May-June) the following year. This combination of methods will eliminate large infestations of RCG, but follow-up (i.e. spot herbicide treatment with a backpack sprayer) will still be required for several years, since some RCG plants will survive or will reinvade the site. Active restoration will be necessary if a remnant seed bank does not exist.

Flooding without tillage: Controlling the hydrology of the site to lengthen the time an area spends totally submerged may be a viable control strategy if you have control over the hydroperiod of your site. Local experiments are underway using this technique by the USFWS at the Sandy River Delta and by the Port of Portland at the Vanport wetlands along the Columbia River. Other treatments will be necessary along the edges of the flooded zone.

Prescribed Fire

Burning generally does not kill mature RCG, and similar to occasional mowing, actually appears to stimulate additional stem production unless the fire burns through the entire RCG sod layer down to the mineral soil (which in turn, may create other problems). In most cases, RCG remains green long into the season, and so does not burn very hot. In the PNW, prescribed fire can only occur in the fall, and burning RCG in fall does little to control it. Herbicide treatment prior to burning can facilitate a prescribed fire, especially outside of typical "fire seasons." Prescribed fire can however, be used as a pretreatment to tillage, shade cloth, or prior to herbicide application for good results, since the fire will remove the aboveground dead litter and standing vegetation. Burning for several years in a row is generally not possible because of lack of fine fuels after the first-year burn.

Solarization, Shade Cloth, & Mulching:
Solarization (essentially baking under clear or black plastic) or the use of a thick woven geotextile shade cloth can be used to eliminate RCG. In dense areas of patchy RCG growth this method can provide specific, targeted control photograph 8). In areas where RCG is mixed-in with desirable species, the kill of those desirable species may or may not be an option. Also, the use of certain materials for this method depends on your overall management goals. There are reports from the Puget Sound region of good RCG control by using several layers of cardboard covered by 4 to 6 inches of wood mulch. The addition of these materials into your site may or may not be acceptable. Excellent control of RCG can also be accomplished by using a thick woven plastic fabric (Mirafi(r) or Amoco(r) brands), held in place by 7-inch gutter spikes and washers and duck-bill tree anchors. The fabric is kept in place for over one year (over an entire growing season), even under inundation. This method will kill all plants under the cloth. Revegetation or reseeding is generally necessary with this method. Shade cloth is initially expensive (approximately $400 per 12 ft x 350 ft roll), but can be reused several times, and this method does not require follow-up visits during treatment. Mowing prior to the installation of shade cloths greatly facilitates installation. Small patches can likely be treated using black plastic bags, if they are kept in place for the entire duration, the edges are tacked-down firmly, and the bags do not shred.

Grazing
Grazing alone does not control RCG. It is likely that much of the RCG present in the PNW today comes from European cultivars planted purposefully for pasture, so grazing has little to no negative impact on the growth and survivorship of RCG. Cattle prefer RCG when stems and leaves are young and succulent, but do not prefer it once stems become old and tough. Goats and sheep will graze on RCG. Grazing can be combined with another treatment method (followed by tillage, herbicide, shade cloth), for good control.

Biological Control
There are no known biological control agents for RCG.

Chemical Control Methods
RCG can be successfully controlled by the proper use of herbicide. Small stands or clumps of RCG can be effectively killed with one application, but large infestations will likely require several applications over several years to be effective. Since RCG frequently grows in wet areas, only aquaticapproved herbicides are allowed in many situations. As with all herbicide use, be sure to read and follow all label instructions and to abide by all state regulations. Glyphosate (Rodeo(r), Aquamaster(r), or Glypro(r) among others) applied in a 2% solution (1.08% active ingredient (a.i.)) with a nonionic surfactant works well to kill RCG. Glyphosate is a non-selective herbicide that kills or injures nearly all plant species. Glyphosate is also available in many other formulations (e.g. RoundUp(r)). These work well to kill RCG, but are not labeled for aquatic use, so be aware of the areas where you plan on applying herbicide. Sethoxydim (Vantage(r)) is a grass-specific herbicide that has been used to kill RCG with some success in the PNW, but it is also not labeled for aquatic use.

Always follow all herbicide label instructions and directions! Non-target organisms that come in contact with the herbicide may be injured or killed. If you have questions about herbicide choice or whether your use is legal, please contact your state Department of Agriculture or local extension agent. Depending on the size and distribution of your infestation, the herbicide can be foliar-applied using a dripless wick applicator, backpack sprayer, or boom sprayer (please visit http://tncweeds.ucdavis.edu for a full discussion of herbicide application tools). Herbicide should be applied to foliage during the growing season. Application in the PNW can occur in mid-summer (just prior to summertime dormancy) or preferably in late fall (just prior to frost and wintertime dieback). It is recommended to apply herbicide at these times, since it is speculated that these are the times of year when RCG is most actively translocating carbohydrates (along with the herbicide) down into the root system.

You may also combine an herbicide treatment with another control treatment for good results. First, eliminate the aboveground dead litter by mowing or burning, then allow the RCG stems and leaves to regrow to boot height. This helps obtain better herbicide coverage and reduce total herbicide use, since you are spraying only living green RCG that is 12" tall vs. 6' tall stems mixed with old dead leaves. Follow-up monitoring and treatment is necessary for several years to ensure complete kill.

Restoration/Competition
Planting fast-growing shrubs or trees may eventually eliminate RCG since it is intolerant of year-round shade, but depending on your management goals and objectives, this may not be a viable option. In the Puget Sound region where forested wetlands are common, planting native evergreen trees (Pseudotsuga menziesii, Picea sitchensis) may be desirable and can successfully shade-out and eliminate RCG. One way to add conifers into a RCG dominated system is to cut holes into large downed woody debris, and plant the conifer seedling into that hole. If planting trees directly into the RCG wetland, the trees will do better if the RCG is kept mowed, or if the trees are planted on top of soil mounds.

In the Willamette Valley however, native wet prairies did not have a large conifer component, and the addition of coniferous trees into this system may be undesirable. Because RCG can survive under the deciduous canopy of cottonwoods (Populus trichocarpa) and Oregon ash (Fraxinus latifolia), the planting of these trees and other native shrubs alone (Spiraea douglasii, Sambucus racemosa) are not be likely to be successful at fully eliminating RCG. However, the City of Portland Bureau of Environmental Services reports success with planting high-density cottonwood or alder. Where they have reached a closed canopy by year five, they report almost near eradication of RCG. While this is not true eradication, it is enough to allow success in moving the site into an artificial conifer succession stage and to try to establish some native forbs. Creating a dense herb layer may be able to exclude RCG, once it is firmly established. Native grasses, sedges and rushes such as Beckmannia syzigachne, Eleocharis palustris, Carex densa, C. feta, C. unilateris, Juncus oxymeris, Deschampsia caespitosa, and Agrostis exarata, along with a native forb component (Myosotis laxa, Plagiobothrys figuratus, Veronica scutellata) may work to exclude RCG in some situations in the Willamette Valley. Further, a complex herbaceous canopy can work to prevent RCG seed germination.

The likeliest scenario for successful exclusion of RCG using native species is habitats that are marginalfor RCG in the first place, i.e. those that are submerged for much of the year. In the transitional zone between the emergent plant community and the upland, where we find the most difficulty with RCG in the Portland Metro region, native sedges, grasses and other emergent species have difficulty in holding their own against

RCG. The City of Portland Bureau of Environmental Services suggests those areas are most easily managed by planting either one of the two following strategies:

1) live stakes- living branch sections from adapted trees/shrubs like willow, dogwood
2) shrub clusters- first scalp the sod off the top of an area about 5' by 3', then plant a cluster of shrubs- we generally use 10 for each cluster, then mulch well with a coarse grained mulch which will keep RCG from coming up from the bottom but also prevent broadleaf weed invasion from the top.

Best Management Practice Recommendation

The best management approach to use will depend on your overall management goals and objectives, the size, distribution and location of your RCG infestation(s), your capability and willingness to use herbicides (or not), and your available resources (staff and volunteer time, money, equipment, etc). The following recommendations are not necessarily the best management methods for every situation, nor are they presented in an order of preference. The methods listed below have however, been used with some success in the PNW. Also, every method will require follow-up monitoring and treatment (including replanting native species if necessary) to ensure the long-term success of your treatments.

Scattered individual plants or small patches in healthy native vegetation
1. Dig out using a shovel
2. Spot-spray or wick with herbicide
3. Spot flame with a propane torch (only works for seedlings or young individuals)

Distinct patches of RCG within a matrix of native vegetation
1. Dig out using a shovel (depends on size)
2. Cover with shade cloth (may be preceded by mowing)
3. Mow (to eliminate seeds), then spot-spray or wick with herbicide
4. Spot-spray or wick with herbicide

Large patches (up to several acres) of RCG with scattered native vegetation (Which method you choose will depend on how much you want to keep your native vegetation)
1. Mow then cover with shade cloth
2. Mow then herbicide (wick, spot-spray or boom)
3. Herbicide using appropriate application technique
4. Spot-burn then spot-spray regrowth
5. Cover with shade cloth (may be preceded by a mow treatment)

Large (hundreds of acres) monocultures of RCG
1. Mow using large mower, herbicide spray using boom sprayer
2. Prescribed burn, then herbicide spray using boom sprayer
3. Tillage and flooding

Return to List




## Common reed
## *Phragmites australis* (Cav.) Trin. ex Steudel

**SOURCE**: The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is May 5, 2005.

**NATIVE RANGE**: Unclear
**DESCRIPTION:** Common reed, or Phragmites, is a tall, perennial grass that can grow to over 15 feet in height. In North America, both native phragmites (*Phragmites australis* ssp. *americanus* Saltonstall, P.M. Peterson & Soreng) and introduced subspecies are found. Introduced *Phragmites* forms dense stands which include both live stems and standing dead stems from previous year's growth. Leaves are elongate and typically 1-1.5 inches wide at their widest point. Flowers form bushy panicles in late July and August and are usually purple or golden in color. As seeds mature, the panicles begin to look "fluffy" due to the hairs on the seeds and they take on a grey sheen. Below ground, *Phragmites* forms a dense network of roots and rhizomes which can go down several feet in depth. The plant spreads horizontally by sending out rhizome runners which can grow 10 or more feet in a single growing season if conditions are optimal.

Visit the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild Fact Sheet for Common Reed to view a table of characteristics that distinguish native Phragmites from the invasive Phragmites at http://www.nps.gov/plants/alien/fact/phau1.htm or at http://www.invasiveplants.net/invasiveplants/phragmites/morphology.htm.




Photo: Jil M. Swearingen, USDI National Park Service, www.forestryimages.org




Photo: James R. Allison, Georgia Department of Natural Resources, www.forestryimages.org




Photo: James H. Miller, USDA Forest Service, www.forestryimages.org

**ECOLOGICAL THREAT:** Once introduced *Phragmites* invades a site it quickly can take over a marsh community, crowding out native plants, changing marsh hydrology, altering wildlife habitat, and increasing fire potential. Its high biomass blocks light to other plants and occupies all the growing space belowground so plant communities can turn into a *Phragmites* monoculture very quickly. *Phragmites* can spread both by seed dispersal and by vegetative spread via fragments of rhizomes that break off and are transported elsewhere. New populations of the introduced type may appear sparse for the first few years of growth but due to the plant's rapid growth rate, they will typically form a pure stand that chokes out other vegetation very quickly.

Photo: © John M. Randall/
The Nature Conservancy



**DISTRIBUTION IN THE UNITED STATES:** *Phragmites* occurs throughout the lower 48 states and southern Canada. It has been reported to be invasive in natural areas in 18 states including Colorado, Connecticut, Delaware, Georgia, Indiana, Kentucky, Maryland, Michigan, North Carolina, New Hampshire, New Jersey, New York, Ohio, Pennsylvania, Tennessee, Virginia, Vermont, and Wisconsin, and the District of Columbia.

**HABITAT IN THE UNITED STATES:** Tidal and nontidal brackish and freshwater marshes, river edges, shores of lakes and ponds, roadsides, disturbed areas.

**BACKGROUND:** Preserved remains of native *Phragmites* that are 40,000 years old have been found in the southwest indicating that it is a part of the native flora of that region. In coastal areas, preserved rhizome fragments dating back 3000-4000 years have also been found in salt marsh sediments indicating that it is also native to these habitats. Native American uses of *Phragmites* include use of stems for arrow shafts, musical instruments, ceremonial objects, cigarettes, and both leaves and stems for constructing mats. Introduced *Phragmites* is thought to have arrived in North America accidentally, most likely in ballast material in the late 18th or early 19th centuries. It established itself along the Atlantic coast and over the course of the 20th century, spread across the continent. In Europe Phragmites is grown commercially and is used for thatching, fodder for livestock, and cellulose production. It is also declining in parts of Europe which has been of concern to natural resource managers there. Here in the United States it is not used for many purposes.

**BIOLOGY & SPREAD:** While each *Phragmites* plant may produce thousands of seeds annually, seed viability is typically low although there appears to be a great deal of interannual variation in fecundity. Dispersal to new sites is typically by seed except along rivers and shorelines where fragments of rhizomes may be washed down to new sites where they can establish. Along roadsides, rhizomes fragments may also be transported by heavy machinery between sites. At this time, there is no evidence for hybrid native/introduced populations occurring in the field.

**SUGGESTED ALTERNATIVE PLANTS:** Native plant species that are adapted to local conditions should be used in restoration projects and as a substitute for *Phragmites* erosion control practices.

**AUTHORS:**
Dr. Kristin Saltonstall, Adjunct Research Scientist, Horn Point Laboratory University of Maryland Center for Environmental Science, Solomon MD.

**EDITOR:** Jil M. Swearingen, National Park Service, Center for Urban Ecology, Washington DC.

**For more information on common reed, please contact:**
Cornell University, Biological Control of Weeds-Common reed, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Delaware River Invasive Plant Partnership, Invasive Plant Fact Sheets, http://www.paflora.org/DRIPP.html.
Dr. Kristin Saltonstall; Adjunct Research Scientist; Horn Point Laboratory; University of Maryland Center for Environmental Science; ksalton@hpl.umces.edu; Tel: (914) 526-2498
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Ohio Department of Natural Resources, Division of Natural Areas and Preserves, Invasive Plants of Ohio Fact Sheet No. 5, Phragmites australis, http://www.dnr.state.oh.us/dnap/invasive/5reedgrass.htm.
The Bugwood Network, MA-EPPC Invasive Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Northeastern Area, Invasive Plants - Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program Fact Sheet-Common reed; http://www.dcr.virginia.gov/dnh/invlist.htm
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/scott/weed_id/genusindex.htm

**REFERENCES:**
Chambers, R.M., L.A. Meyerson, and K. Saltonstall. 1999. Expansion of Phragmites australis into tidal wetlands of North America. Aquatic Botany 64: 261-273.
Marks, M., B. Lapin, et al. 1994. Phragmites australis (P. communis): Threats, management, and monitoring. Natural Areas Journal 14: 285-294.
Meyerson, L.A., K. Saltonstall, L. Windham, E. Kiviat, and S. Findlay. 2000. A comparison of Phragmites australis in freshwater and brackish marsh environments in North America. Wetlands Ecology and Management 8: 89-103.
Norris, L., J. E. Perry, and K.J. Havens. 2002. A summary of methods for controlling Phragmites australis. Virginia Institute of marine Science Wetlands Program Technical Report No. 02-2.
Saltonstall, K. 2002. Cryptic invasion of a non-native genotype of the common reed, Phragmites australis, into North America. Proceedings of the

National Academy of Sciences USA 99(4): 2445-2449.
Saltonstall, K. 2003. Microsatellite variation within and among North American lineages of Phragmites australis. Molecular Ecology 12(7): 1689-1702.
Saltonstall, K. 2003. Genetic variation among North American populations of Phragmites australis: implications for management. Estuaries 26 (2B):445-452.
Saltonstall, K. 2003. A rapid method for identifying the origin of North American Phragmites populations using RFLP analysis. Wetlands 23(4) 1043-1047.
Saltonstall, K., P.M. Peterson, and R. Soreng. 2004. Recognition of Phragmites australis subsp. americanus (Poaceae: Arundinoideae) in North America: evidence from morphological and genetic analyses. Sida.
Swearingen, J. 2005. Alien Plant Invaders of Natural Areas. Plant Conservation Alliance, Alien Plant Working Group. http://www.nps.gov/plants/alien/list/a.htm
U.S.Department of Agriculture. 2005. Natural Resources Conservation Service Plants Database. http://plants.usda.gov/

**Management and Control Information**

Return to List




## Species Management and Control Information

# Common reed
# *Phragmites australis ssp. australis*

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Phragmites australis - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Careful planning and long-term management can yield varying levels of control of common reed. Because a healthy wetland ecosystem is generally resistant to invasive species, long-term control of common reed depends upon restoration of the health of the ecosystem.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Common reed - http://www.nps.gov/plants/alien/fact/phau1.htm - Author: Sr. Kristin Saltonstall - May 5, 2005.**

Areas with large, established, populations of Phragmites are best restored using herbicides. Other options include mowing and prescribed burning.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Phragmites australis - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Authors: Marianne Marks, Beth Lapin and John Randall - May 12, 1993.**

Management Requirements:
Invasive populations of Phragmites must be managed in order to protect rare plants that it might outcompete, valued animals whose habitat it might dominate and degrade, and healthy ecosystems that it might greatly alter.

Management Programs:
Cultural, mechanical and/or chemical methods can be used to control Phragmites. The factors that are believed responsible for the alarming decreases of Phragmites beds in Europe and Texas include habitat destruction, increased soil nitrate levels, and eutrophication (Boar, Crook and Moss 1989, Ostendorp 1989, Sukopp and Markstein 1989) are not appropriate as management tools in natural areas.

## MANUAL AND MECHANICAL CONTROL:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Phragmites australis - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Cutting, pulling, or mowing can be done in late July and should be repeated for several years. All cut shoots should be carefully removed to prevent resprouting. The placement of black plastic over cut stems has produced some success and burning in combination with herbicide application has also been effective in some situations. Hydrologic controls such as flooding for an extended period during the growing season may also be successful.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet No. 5 - Phragmites australis - http://www.dnr.state.oh.us/dnap/invasive/5reedgrass.htm - January 3, 2003.**

Cutting, pulling or mowing can be done in late July and should be repeated for several years. All cut shoots should be carefully removed to prevent re-sprouting. The placement of black plastic over cut stems has had some success and burning in combination with herbicide application has also been effective in some situations. Hydrologic controls such as flooding for an extended period during the growing season may also be successful.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Common reed - http://www.nps.gov/plants/alien/fact/phau1.htm - Author: Sr. Kristin Saltonstall - May 5, 2005.**

This type of control (e.g., repeated mowing) may be effective at slowing the spread of established stands but is unlikely to kill the plant. Excavation of sediments may also be effective at control but if small fragments of root are left in the soil, they may lead to reestablishment.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Phragmites australis - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Authors: Marianne Marks, Beth Lapin and John Randall - May 12, 1993.**

CUTTING: Cutting has been used successfully to control Phragmites. Since it is a grass, cutting several times during a season, at the wrong times, may increase stand density (Osterbrock 1984). However, if cut just before the end of July, most of the food reserves produced that season are removed with the aerial portion of the plant, reducing the plant's vigor. This regime may eliminate a colony if carried out annually for several years. Care must be taken to remove cut shoots to prevent their sprouting and forming stolons (Osterbrock 1984). In the Arcola Creek Preserve in Ohio, cutting reduced the vigor of the Phragmites colony. Also in Ohio, at Morgan Swamp, cutting began in mid to end of July (before tassel set) in 1989 around a gas well in a freshwater wetland (Seidel, pers. comm. 1991). The preferred tool was an old-fashioned hedge trimmer with an 8" flat blade with serrations manufactured by Union Fork and Hoe. The trimmers worked better than loppers and were safer than sickles; a circular blade on a weed whacker was also used and proved to be faster and good for staff but it was more dangerous for volunteers and detracted from the atmosphere of the work-day (Huffman, pers. comm. 1992).

Small patches (10' x 50') in a New York freshwater system were cut at the end of July or the beginning of August for two successive years with positive results (Schneider, pers. comm. 1990). The hand-cut material was removed from the site and thrown on a brush pile (unfortunately it was located too close to the water and returned to the system).

Massachusetts Audubon staff have cut the perimeter around a 0.25 acre Phragmites patch at the end of July since 1986 in a freshwater wetland at Daniel Webster Preserve in Marshfield, Massachussetts (Anderson, pers. comm. 1992). They have monitored their success in keeping it from spreading by using a map and hand compass.

Stands of Phragmites of less than 1 acre in extent that block views in Everglades National Park are cut just before the onset of the rainy season. The rise in water elevation from the rains that follow stresses the roots of the plant. This works to a degree but Phragmites returns (Dowlen, pers. comm. 1985).

In Quincy, Mass., the town used small Bobcats with lawnmower clippers mounted on the buckets with a flexible cable to cut an area with 75% cover of Phragmites and 20-25' of muck (Wheelwright, pers. comm. 1991; Dobberteen pers. comm. 1991). Cutting this 10-acre plot three times during the summer (April, June, August) cost $150K. The cut material was stockpiled nearby where it was to be burned in the winter when it was washed away in a severe storm. In winter 1992, the town plans to open the tide gate and allow flushing to prevent further return of Phragmites. Results are not yet known.

Cutting culms to 6" followed by addition of rock salt on a 10' x 10' patch appeared to have reduced the height and density of Phragmites in a salt marsh in Greenwich, CT (Jontos and Allan 1984). Continued observations indicated that this trend appeared to continue (Jontos, pers. comm. 1992).

Cutting an area 25' x 25' to waist height with a hedge clippers and the applying one drop of Roundup with a syringe with a large needle (horse size) into the top of the plant in a brackish- freshwater marsh was begun in Constitution Marsh in New York in 1991 (Keene, pers. comm. 1991). Initial results indicate 90% eradication.

In Connecticut, cutting below the first leaf at the end of July in 1986, 1989, 1990, 1991, and 1992 in a freshwater tidal wetland around the perimeter of a one-acre patch has prevented subsequent expansion of the patch. Monitoring using aerial photos taken at five-year intervals indicated the control success. Cutting was done with hand-held cutters and gas-powered hedge trimmers, which were very efficient. Cut material was removed from the site and allowed to decompose on upland areas. In a second area, similar efforts in a calcareous wetland 1990-1992 were monitored by placing red survey wires around the perimeter of the patch. Preliminary observations indicate a cessation of Phragmites expansion.

In Europe, Weisner and Graneli (1989) found that oxygen transport was reduced by cutting the culms above and below the water surface;cutting below the water in June almost totally inhibited regrowth of shoots the following summer, while cutting above water reduced regrowth of shoots. Cutting in August did not reduce growth the following summer. Cutting in sandy substrates was minimally effective, while cutting on calcareous muds caused decreases in oxygen levels.

GRAZING, DREDGING, AND DRAINING: Grazing, dredging, and draining are other methods that have often been used to reduce stand vigor (Howard, Rhodes and Simmers 1978). However, draining and dredging are not appropriate for use on most preserves (Osterbrock, 1984).

Grazing may trample the rhizomes and reduce vigor but the results are limited (Cross and Fleming 1989). Van Deursen and Drost (1990) found that cattle consumed 67-98% of above-ground biomass; in a four year study, they found that reed populations may reach new equilibria under grazing regimes.

MANIPULATION OF WATER LEVEL AND SALINITY: A self-regulating tide gate which reintroduced saltwater tidal action was used to help restore a diked marsh in Fairfield, Connecticut (Thomas Steinke pers. comm. 1992; Bongiorno et al. 1984). A 1-3 foot reduction in stem height resulted over each of three years . In addition to reduced height, plant density declined dramatically from 11.3 plants m-2 in 1980 to 3.3 plants/ m-2 the following year. In following years, Phragmites continued to decline, although less dramatically. In addition to the decreased height and density of the Phragmites stands, typical marsh flora including SALICORNIA, DISTICHILIS, SPARTINA ALTERNIFLORA Loisel. and S. PATENS (Aiton) Muhl. returned. Depending on topography and elevation, Phragmites was eliminated in large areas and continues to remain short and sparse in other areas through 1992. Hence, reintroduced tidal action and salinity can reduce Phragmites vigor and restore the community's integrity. This has been implemented successfully in other degraded former salt marshes in Connecticut (Rozsa, pers. comm. 1992).

Flooding can be used to control Phragmites when 3 feet of water covers the rhizome for an extended period during the growing season, usually four months (Beall 1984). However, many areas can not be flooded to such depths. Furthermore, flooding could destroy the communities or plants targeted for protection.

Open Marsh Water Management (OMWM) has been used as a method to control Phragmites. Plugging of ditches and addition of culverts to raise the soil salinities appears to have caused Phragmites die-back over the last four growing seasons at Fireplace Neck, New York (Niniviaggi, pers. comm. 1991; Rozsa, pers. comm. 1992).

Hellings and Gallagher (1992) found that Phragmites was negatively impacted by increasing salinity and increased flooding. They also found that cutting and subsequent flooding also reduced growth and survival in outdoor experiments. They suggest that Phragmites may be controlled by increasing flooding and salinity levels. Matoh, Matsushita and Takahashi (1988) also found reduction in vigor with increased salinity. However death apparently occurred only when cutting was combined with brackish flooding (Hellings and Gallagher 1992).

In Europe, episodic freshwater flooding occurring early in the growing season has been suggested as one of the reasons for reed population declines (Ostendorp 1991). McKee et al. (1989) investigated root metabolic changes due to freshwater flooding and labelled Phragmites as a flood-tolerant species.

Also see Chincoteague NWR under CHEMICALS, Wertheim NWR under BURNING, and Town of Quincy under CUTTING for additional references.

MOWING, DISKING, AND PULLING: Beall (1984) discourages mowing and disking. Mowing only affects the above ground portion of the plant, so mowing would have to occur annually. To remove the rhizome, disking could be employed. However, discing could potentially result in an increase of Phragmites since pieces of the rhizome can produce new plants. Cross and Fleming (1989) describe successful mowing regimes of several year duration during the summer (August and September) and discing in summer or fall.

In Cape May Meadows, New Jersey, a brackish to freshwater non- tidal sandy area, an attempt was made to remove rhizomes by pulling to a depth of three feet (Johnson, pers. comm. 1991). This resulted in a very sparse Phragmites stand the following year. However it was very labor-intensive (using 130 people- hours to cover a 50 ft2 patch) and could be applied best to sandy soils.

In a private yard, Phragmites was mowed and a thin layer of soil and grass seed were added. This was mowed weekly over the course of the summer. In the second summer shoots of Phragmites occurred around the edges. The rhizomes were decomposing after this treatment (M. Ailes, pers. comm. 1992).

PLASTIC: Clear plastic six-mil thick, 12 x 17 m, weighing 51.8 kg, was carried into a North Carolina marsh by air and held in place by sandbags (Boone et al. 1987, 1988). Plants were initially cut to 6-8" with a hand-pushed bush hog (Boone, pers. comm. 1991) or a weedeater with blade, with an area of 20 x 20 m taking several days to cut. The cut material was left and the plastic put over the area. The high temperatures under the plastic caused die-off of Phragmites in 3-4 days. After 8-10 weeks, the plastic deteriorated. The rhizomes appeared to have died back, but the project was of short duration and the results were not monitored the following year (Boone, pers. comm. 1991). Turner (pers. comm. 1992) noted that follow-ups in subsequent years indicated Phragmites returned but not as densely. Plastic management in each 12 x 12 m plot took an average of 53 hours, compared with 17 hours to cut and three hours to burn (Boone et al. 1987).

Clear plastic in two narrow swaths (70 m x 20 m) was placed along the edge of a tidal brackish pond after hand-cutting the Phragmites at the end of July 1991 (Anderson, pers. comm. 1992). One plot, in total sun, had a complete kill of Phragmites in 10 days, while the plot in partial shade had a partial kill. It is unknown how the plastic was kept in place or what was done with the cut material.

Clear and black plastic were used on 50' circular areas at Constitution Marsh in New York in 1990 and 1991 (Keene, pers. comm. 1991). Although there was difficulty due to tidal influence, the plastic was weighted down with rocks and appeared to kill what is under it. Runners along the edge were treated with a syringe application of Roundup in August. In November 1991, a hole cut in the middle of the black plastic provided the opportunity for cattail shoots to germinate. After the first year there was viable Phragmites in the areas covered. It appeared that the black plastic was more effective, due to the higher heat levels attained (Rod, pers. comm. 1992).

## PRESCRIBED FIRE:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Common reed - http://www.nps.gov/plants/alien/fact/phau1.htm - Author: Sr. Kristin Saltonstall - May 5, 2005.**

Prescribed burning after the plant has flowered, either alone or in combination with herbicide treatment, may also be effective. Burning after herbicide treatment also reduces standing dead stem and litter biomass which may help to encourage germination of native plants in the following growing season. Plants should not be burned in the spring or summer before flowering as this may stimulate growth.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Phragmites australis - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Authors: Marianne Marks, Beth Lapin and John Randall -**

**May 12, 1993.**

Prescribed burning does not reduce the growing ability of Phragmites unless root burn occurs. Root burn seldom occurs, however, because the rhizomes are usually covered by a layer of soil, mud and/or water. Fires in Phragmites stands are dangerous because this species can cause spot-fires over 100 feet away (Beall 1984). Burning does remove accumulated Phragmites leaf litter, giving the seeds of other species area to germinate. Prescribed burning has been used with success after chemical treatment for this purpose at The Brigantine National Wildlife Refuge, NJ (Beall 1984) and in Delaware (Lehman, pers. comm. 1992). Occasional burning has been used in Delaware in conjunction with intensive spraying and water level management. This helps remove old canes and allows other vegetation to grow (Daly, pers. comm. 1991)

At Wallops Island, Virginia, a small (100' x 400') brackish to saline to dry wetland was burned November 1990 to control Phragmites (M. Ailes, pers. comm. 1992). A variety of other species appeared in the year following the burn but they appeared leggy while the Phragmites remained vigorous. A second winter burn is planned and monitoring of transects will continue (there are no pre-treatment data).

At Wertheim National Wildlife Refuge in New York, a 20-30 acre freshwater impoundment was drained in the fall of 1989, burned the following winter and then reflooded (Parris, pers. comm. 1991). Phragmites was eliminated from the half of the marsh that was treated and the area remained free of the grass through 1992.

According to Cross and Fleming (1989), late summer burns may be effective, but winter and spring burning may in fact increase the densities of spring crops. Thompson and Shay (1985) performed experimental burn treatments on Delta Marsh, Manitoba. They found that spring, summer and fall burns resulted in higher total shoot densities and lower mean shoot weights than on controls primarily as a result of greater densities of shorter, thinner vegetative shoots. Shoot biomass was greater in spring-burned and fall-burned plots than in control areas but less on summer-burned plots. They also found that below-ground production increased following spring and fall burns but not following summer burns. The increase in light availability following burns generally appears to benefit Phragmites. A variety of understory responses to these burns was noted. For example, summer burns increased species diversity, richness, and evenness, although certain species declined (Thompson and Shay 1985).

In Connecticut late spring burns followed by manual flooding with salt water was successful in reducing Phragmites height and density (Steinke, pers. comm. 1992). After three years, the fuel load was exhausted; the process was very expensive and self- regulating tide gates were installed instead (see MANIPULATION OF WATER LEVEL AND SALINITY).

In Europe, experimental removal of litter in winter resulted in doubling the above-ground biomass (Graneli 1989). Increased light availability at the soil surface and aeration of the soil around the rhizomes may have been responsible for this increase. Burning in the winter in an experimental field caused little damage, while burning during the emergence period led to the death of the majority of Phragmites shoots (van der Toorn and Mook 1982).

## BIOCONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Common reed - http://www.nps.gov/plants/alien/fact/phau1.htm - Author: Sr. Kristin Saltonstall - May 5, 2005.**

At this time no means of biological control are available in the United States for treating Phragmites infestations

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Phragmites australis - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Authors: Marianne Marks, Beth Lapin and John Randall - May 12, 1993.**

Biological control does not appear to be an option at this time. No organisms which significantly damage Phragmites australis but do not feed on other plant species have been identified. Naturally occurring parasites have not proven to be successful controls (Tscharntke 1988, Mook and van der Toorn 1982, van der Toorn and Mook 1982). In addition, some of the arthropods that feed on Phragmites are killed by winter fires and thus would likely be eliminated from the systems where prescribed fires are used. Coots, nutria, and muskrats may feed on Phragmites but appear to have limited impacts on its populations (Cross and Fleming 1989).

**USDA Forest Service - Biological Control of Invasive Plants in the Eastern United States - http://www.invasive.org/eastern/biocontrol/9CommonReed.html - Publication FHTET-2002-04 - Authors: R. Van Driesche, et al., 2002.**

No deliberate introductions of biological control agents have been made. The diversity of accidentally introduced Phragmites herbivores is highest closest to New York City (Blossey and Eichiner, unpub.). This suggests that a major area for the introduction of arthropods is the harbor. Various introduced species associated with Phragmites appear to be spreading from New York City along highways, rivers, and the coastline.

Suppression of Target Weed: No work on evaluating the effects of these European herbivores on Phragmites has yet been done in North America. However, the recent discovery of several such species in the northeast provides an opportunity to measure the influence of these organisms on Phragmites performance.

## CHEMICAL CONTROL*:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Phragmites australis - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April 2002.**

Herbicide application with glysophate is most effective in the early fall, after flowering; because common reed often grows in or near water, only formulations approved for aquatic use (Rodeo) should be utilized. Applications will need to be made at least two years in a row. Fusilade DX®, a grass specific herbicide, can be applied in non-aquatic areas. Methods of application will depend on the associated plant community but may

include aerial spraying, hand-held or backpack sprayers, and hand wicking. Herbicide use in combination with burning has generally proven to be the most effective means of control, and results in minimal disturbance to wetlands. Only a biodegradable herbicide that is licensed for use in wetlands and is non-toxic to animals can be used.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet No. 5 - Phragmites australis - http://www.dnr.state.oh.us/dnap/invasive/5reedgrass.htm - January 3, 2003.**

Herbicide application with Accord®, Rodeo® or Glypro® is most effective in the early fall, after tasseling, and should be applied at least two years in a row. Fusilade DX®, a grass specific herbicide can be applied in nonaquatic areas. Methods of application will depend on the associated plant community but may include aerial spraying, hand-held or backpack sprayers and hand-wicking.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Common reed - http://www.nps.gov/plants/alien/fact/phau1.htm - Author: Sr. Kristin Saltonstall - May 5, 2005.**

Glyphosate-based herbicides (e.g., Rodeo) are the most effective control method for established populations. If a population can be controlled soon after it has established chances of success are much higher because the below-ground rhizome network will not be as extensive. Herbicides are best applied in late summer/early fall after the plant has flowered either as a cut stump treatment or as a foliar spray. It is often necessary to do repeated treatments for several years to prevent any surviving rhizomes from resprouting. When applying herbicides in or around water or wetlands, be sure to use products labeled for that purpose to avoid harm to aquatic organisms.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Phragmites australis - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Authors: Marianne Marks, Beth Lapin and John Randall - May 12, 1993.**

RodeoTM, a water solution of the isopropylamine salt of glyphosate is commonly used for Phragmites control. This herbicide is not, however, selective and will kill grasses and broadleaved plants alike. Toxicity tests indicate that it is virtually non-toxic to all aquatic animals tested. It should be noted that many of these tests were performed by or for Monsanto, the company which manufactures Rodeo. Bioconcentration values for glyphosate in fish tissues were insignificant. Glyphosate biodegrades quickly and completely in the environment into natural products including carbon dioxide, nitrogen, phosphate and water. Finally, since glyphosate does not volatilize, it will not vaporize from a treated site and move to a non-target area (Brandt 1983; Comes, Bruns and Kelly 1976; Folmar, Sanders and Julin 1979; Monsanto 1985).

Rodeo must be mixed with water and a surfactant which allows it to stick to and subsequently be absorbed by the plant (Beall 1984). Instructions for application, amounts needed per acre, the approved surfactants and ratios for mixing, are on the Rodeo label. Glyphosate must be mixed with clean or, if possible, distilled water because it binds tightly to sediments and is thus rendered non-toxic to plants (Lefor, pers. Comm. 1992). This limits its effectiveness but also may help prevent it from acting on plants that were not originally targeted. Rodeo should not by applied in windy conditions, as the spray will drift (I. Ailes, pers. comm. 1985). It also should not be applied if rain is forecast within 12 hours because it will wash away before it has a chance to act (Daly 1984). Application rates may vary but, as one example, effective control of Phragmites in a Delaware marsh was achieved with 4 pints/acre of concentrate (Lehman, pers. comm. 1992).

Application of Rodeo must take place after the tasseling stage when the plant is supplying nutrients to the rhizome. At this time, when Rodeo is sprayed onto the foliage of aquatic weeds, it translocates into the roots. Rodeo interferes with essential plant growth processes, causing gradual wilting, yellowing, browning and deterioration of the plant. Studies on tasseling at the Augustine Tidal area, in Port Penn Delaware, indicated that tasseling in a stand is never 100% but that it is possible to spray when 94% of the plants are tasseling. In dense stands, subdominant plants are protected by the thick canopy and thus may not receive adequate herbicide. For these reasons, touch up work will be necessary (Lehman 1984).

At Brigantine National Wildlife Refuge, Rodeo was applied aerially after the plants tasseled in late August. The application resulted in a 90% success. The following February, a fast moving prescribed burn was carried out to remove litter, exposing the seed bed for re-establishment of marsh vegetation. However funding was not available for several years and Phragmites has returned to 90% of the previously treated areas (Beall, pers. comm. 1991). Treatment was resumed in fall 1991.

In September, 1983, at the Prime Hook Wildlife Refuge in Delaware, 500 acres of freshwater impoundments were sprayed with Rodeo from a helicopter for Phragmites control. The plants yellowed within 10 days. The following May aerial and ground evaluations of the sprayed area revealed a 98% kill of Phragmites (Daly 1984). In addition to applying herbicide, Prime Hook manipulates water levels with a stop log to stress Phragmites; winter water levels are held at an elevation of 2.8' msl until June, when water would otherwise be held at 2.2 msl. The combined spraying and water management approach was successful and many aquatic plants returned. A regime of spraying in August-September for two years followed by flooding has been used through 1991 (Daly, pers. comm. 1991). Annual costs of Phragmites control are $20K annual at Prime Hook (1,000 acres) and $3K at Bombay Hook (20-60 acres); monitoring costs, which include reading vegetation transects for species presence and density each September are not included in the cost.

Aerial spraying has been used since 1983 in many Delaware state wildlife refuges (Lehman, pers. comm. 1992). Using Rodeo, the state sprays freshwater and brackish impoundments, brackish marshes, and salt marshes from early September to early October; this is combined with winter burns between the first and second year of spraying. Areas will be spot-treated whenever needed after that. The herbicide treatments consist of 4 pints/acre the first year and 2 pints/acre the second, with an average cost of $65/acre. The state is involved with cost-sharing programs with private landowners where the state pays half the spraying cost with a willing owner. Desirable native vegetation usually returns after spraying; no revegetation is done. Occasionally become open mud flats that are eventually repopulated by Phragmites.

At Chincoteague National Wildlife refuge, an aerial spraying program initiated in 1986 in an 18-mile long freshwater impoundment was terminated due to budget cuts. Phragmites quickly reclaimed the area, estimated to be 100-150 acres total in small scattered stands (I. Ailes, pers. comm. 1991). In September 1991, spraying with Rodeo began again; it is expected that the entire area will be sprayed again in 1992, and that small areas of re-growth will be sprayed in 1993. Because the area is impounded, the water level usually is lower in the spring, which helps prevent Phragmites regrowth.

Herbicides are used at Tinicum Environmental Center, because other control options are limited. Unplanned burns do occur, but prescribed burns

are not allowed due to the proximity to the highway and airport. Tinicum was recently granted $2M to restore a 18-acre site. Here they will be altering the elevation of the marsh, seeding with native plants, and monitoring the results (Nugent, pers. comm. 1991).

At Parker National Wildlife Refuge, an aerial spraying program (annual budget $5K) for 50 acres of a 100-acre freshwater impoundment began in mid-August 1991. A winter burn is anticipated and a second year of spraying planned. Results will be monitored by using aerial photos to delineate the boundaries of the Phragmites clones. A nearby tower also provides a suitable viewing point to observe progress (Healey, pers. comm. 1992).

In more fragile situations where Phragmites is threatening a rare plant or community, aerial spray techniques are inappropriate because such large-scale application could kill the community that the entire operation was designed to protect. Glyphosate can be applied to specific plants and areas by hand with a backpack sprayer. Wayne Klockner of The Nature Conservancy's Maryland Field Office has been successful in eliminating most Phragmites at the Nassawango preserve by applying glyphosate by hand with a backpack sprayer (Klockner, pers. comm. 1985). The control program there began in 1983; actual spraying is conducted along the power line ROW by Delmarva Power (Droege, pers. comm. 1991). Delmarva Power generally sprays with trucks, backpacks or helicopter, depending on the accessibility of the area and presence of rare plants nearby (Johnstone, pers. comm. 1991). They use Rodeo in tidal areas, and AccordTM (another glyphosate product) in non-tidal areas from mid-August to mid-October, when the plants are going to seed. They spray intensively the first year, and conduct touch-up spraying the second year which eliminates 90-95% of the plants. They then return every three years to eliminate any new plants. They do not spray if the plants are not tasselling and are short.

Rodeo was used at Cape May Meadows in 1989, 1990, and 1991. It was applied with a 30 gallon gas-powered tank with spray nozzle mounted on a truck, Indian pump sprayers, 2.5 gallon hand-held sprayers, and wick applicators (Johnson, pers. comm. 1991). This appeared to kill most, if not all, of the treated Phragmites in this 20-acre area; plants found in the area following treatment were shorter and the stand was less dense (determined visually). However the dead stalks remained and blocked views from the trail.

In Connecticut a 5m x 23 m patch of Phragmites has been treated with a hand-held spray of Rodeo (1988 and 1989) and Roundup (1990 and 1991) for four years in late August-early September. The Phragmites is shorter and less dense at the site but it is still present (Lapin pers. obs.). Actions to supplement and enhance herbicide applications including the removal of tassels (1991) and removal of dead stalks (planned 1992), have been and will be taken. Other chemicals have been used on Phragmites and are described in Cross and Fleming (1989).

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

## OTHER CONSIDERATIONS*:

**The Nature Conservancy - Invasive Species Initiative - Techniques from TNC Stewards for the eradication of Lythrum salicaria (purple loosestrife) and Phragmites australis (common reed/Phrag) in wetlands - http://tncweeds.ucdavis.edu/esadocs/phraaust.html - Editor: Mandy Tu - 2000**

This past summer (2000), Betsy Lyman, the Assistant Director of Science & Stewardship for TNC's Pennsylvania Field Office, sent out a call in both Stew News (an in-house TNC publication) and on TNC's Invasive Species Listserve requesting control information for Lythrum salicaria and Phragmites australis. There was a large response to these questions, and we gathered this information, and lightly edited it. If you have either of these weeds on your preserve, you may benefit from reading about others' control experiences.

**Contents:**

I. Betsy's original inquiry and questions.

II. Personal accounts/Case studies
- A. Excellent control of P. australis. L. salicaria eradication still in progress. (Indiana -- C. O'Leary)
- B. Great success in controlling P. australis. Control of L. salicaria promising after 2 years. (Indiana -- M. Norris)
- C. Excellent, but labor-intensive, control of P. australis. Potential successful control of L. salicaria. (New Jersey -- M. Morris)
- D. Effective control of L. salicaria. (Washington -- M. Summers)

III. Methodologies
- A. Application of herbicide to P. australis and L. salicaria. (Michigan -- J. McGowan-Stinski, D. Borneman, K. Condict)
- B. Adaptive management for P. australis control. (New York -- B. Patterson)

IV. Suggested references

V. Contact information

**I. Betsy's original inquiry and questions**

Many Lythrum salicaria questions (Pennsylvania)

Pennsylvania has a fen complex which contains federally and state-listed plants and reptiles. We have both Phragmites australis and Lythrum

salicaria (purple loosestrife) in several of these fens. We have used groups of volunteers to cut the offending plants (for Phragmites, we cut in mid-June and mid-September; for loosestrife, we cut in late July/early August when it is blooming). However, the US FWS does not like the idea of hordes of volunteers trampling through the fens, since they could easily harm the nests of the endangered turtle. So we are looking into using herbicides-i.e. Rodeo (glyphosate).

1) Are there any studies on the effects of glyphosate applications on turtles?

2) What methods (manual, herbicides, etc.) have you used that were effective in controlling the weeds, but which have not negatively impacted the native vegetation? What worked best?

3) Have you done your control work stepwise (i.e. a little bit each year) or have you done the herbiciding all at once?

4) Have you followed up with any planting of native plants?

**IIA. Case study: Control efforts in Indiana**

We have worked with those two wicked invasive plants in fens and other wetlands. We have had great success with killing Phragmites australis. We use 1.5% Rodeo applied from backpack sprayers with 5 foot wand extensions. (The long wands are necessary to reach the tops of the plant). Our best results have been after late-season spraying, close to, but before, the plants senesce. In Indiana this is the first 3 weeks of September. This technique has been used in large monotypic stands (5-20 acres) where the understory is degraded (mostly Phragmites duff) and we have measured over 97% mortality after the first year of spraying. We then burn the duff the next spring to make follow up easier and to allow the native seed bank to germinate.

In areas where the Phragmites is occasional and intermingled with the natives, we use our best discretion - either carefully spray small stands or hand-cut the stems and drip 25% Rodeo into the stump. Hand-cutting is very time consuming if the stands are really big and is less effective (50-75% mortality), but there is no overspray. We have found that workdays with a lot of hands makes it less tedious and more practical to achieve results. If you decide to spray, which is much more time efficient, the sprayer should be experienced, and overspray can be controlled pretty well.

We have been less successful with purple loosestrife (Lythrum salicaria). My opinion is that hand-cutting and pulling are a complete waste of time and lead to great frustration. Foliar spray with 1.5-3.0% Rodeo is our current technique. In two years of intensive work, we have pushed purple loosestrife from a dominant plant to an occasional plant, but eradication is still many years away. This is due to the incredible seed bank. These plants are tough and sometime require a second application of herbicide to kill them. We generally patrol a site three times in the summer as they do not all flower at the same time. So, we spray once in early July, early August, and early September. (the two loosestrife eating insects are also an option. I am personally opposed to introducing another exotic organism to our systems, but many in TNC think it is the only option. I can send you a contact if you like).

In general, I have found fens to be tricky places to use herbicide - especially foliar application. Grasses and sedges are particularly sensitive and you can get large brown-outs if you are not careful. As I said above, dense stands of Phragmites are better targets for foliar spray as the overspray is less important. If the Phragmites is intermingled with native species, and you can get the help, hand-cutting may be the way to go. I think you have to bite the bullet with purple loosestrife and accept a small amount of overspray in order to control it. We have found that oversprayed areas are re-colonized by natives pretty quickly. As a colleague always reminds me, doing nothing is a management strategy also - in the case of purple loosestrife means the end of your natural area - and this may be the most important thing you can impress on folks.

I have tried to locate info on herbicides and turtles with no luck. A couple of ideas that you may have already thought of : (1) does FWS know where the preferred nesting grounds are? These are often the high and dry parts of the fen complex and can be avoided, (2) These species are usually in low densities, so your chance of spraying individuals directly is very low, (3) I have yet to encounter a turtle in a stand of Phragmites - it is too difficult to get into.

Chip O'Leary, Restoration Coordinator, TNC - Indiana Chapter

**IIB. Case study: Control efforts in Indiana**

Here in Indiana, I manage several fens with invasive plant problems. I have never read anything that said glyphosate was bad for turtles. The story is that the surfactant used in RoundUp (glyphosate) is bad for amphibians and other wetland animals, so that is why Rodeo (glyphosate) is used, which is basically RoundUp without the surfactant.

We have been spraying Phragmites australis in the early fall, September through October, when the seed head is fully emerged, with Rodeo at recommended rates with a solo backpack sprayer with a 5-foot wand. If you spray the top leaves of the plant they die really easily within a few weeks and then one moderate follow-up treatment is needed the next year. If you are careful, you rarely get any herbicide on other plants. We treated areas up to 5-6 acres in size and have had great success. The next year native plants come back from the seed bank under the mat of dead vegetation. A follow-up prescribed burn can also help to remove the large mat of dead Phragmites.

Sometimes for Phragmites, we cut the plant a few inches above the ground and drip herbicide into the cut stem at about 25%, and that has about 50% success, and this is helpful in areas you do not want to overspray or are sensitive. I think herbicide is the best control method for Phragmites. It works well and overspray can easily be minimized in thick stands.

For purple loosestrife (Lythrum salicaria) we have been spraying in a similar way when the flowers appear on the plant. Herbiciding is done frequently throughout the late summer and early fall, especially on flowering plants. It will take several years of spraying to make a real impact on the site. If you are careful and use an adjustable spray nozzle you can really minimize overspray. It is tough when you see some browning around the area of natives, but just remember they are generally tougher than exotics, and will come back soon! You can also replant the area with native plugs and that helps out a lot. Loosestrife is tough and there is generally some overspray, but native plants quickly come back. It has a

large seedbank and takes many years to deplete. I would not replant a loosestrife area with natives for awhile, since the seedbank can take several year to deplete.

Some DNR people in Indiana have released the beetle that eats loosestrife, and have seen pretty good success so far. The beetle may help deplete the seed bank quicker than herbiciding. Our herbicide work on TNC property for loosestrife is coming along really well after only two years of spraying.

Mike Norris, Restoration Specialist, Southern Lake Michigan Rim Project

**IIC. Case study: Control efforts in New Jersey**

What methods have you used that have been effective in controlling the weeds, but don't negatively impact the native vegetation?

In northern New Jersey, we have used cut-stem treatment to control both Phragmites and purple loosestrife at several wetland sites. We have found that injecting the cut stems of Phragmites with Rodeo (50%) after the tassel stage (mid-September-October) is very effective. We thought of removing flowering spikes from the site but since staff was limited, we did not follow through with this idea. Later we learned that seeds are often not viable and germination and successful seedling establishment is rare, so we thought it was not worthwhile to remove the flowers.

The cut-stem treatment appears to have killed most of the underground root system because there has been little to no re-sprouting 2 years after control. While this method results in minimal off-target damage, it is very labor intensive. It will take us three years to treat several small infestations of Phragmites on one of our preserves using both volunteers and 4 full-time staff. This method is probably not worthwhile for people who have large infestations.

This year we also began controlling purple loosestrife in an emergent marsh using cut-stem treatment. Stems were cut near the base and Rodeo (30%) was squirted onto cut stems when plants began flowering (July-August). Herbicide was applied only to stems of target plants, thereby reducing exposure to other species. Cut plant material was bagged and removed from the site. Since control was initiated this year, we do not have information about the effectiveness of this method yet.

What is the most effective means of controlling the weeds (manual, pesticide, combination)?

We have found a combination of manual removal (i.e. cutting) followed by pesticide application to be most effective. See answer to #2.

If you're herbiciding, have you done your treatments stepwise or have you done the herbiciding all at once? It would be useful to hear how large the areas are that you are treating.

Whenever possible we have treated the entire infestation at once. In situations where we don't have enough time or staff to treat the whole area we have been working on patches of the infestation in a stepwise manner.

The Phragmites infestations vary in size. We have treated small patches with only several dozen stems to patches 50 X 25 m. This fall we plan to use volunteers to help us finish treating a Phragmites infestation that is approximately 40 X 40 m. Most of the Phragmites infestations are dense monocultures with only a few other native species present.

The area where we treated purple loosestrife was approximately 0.5 acres. However, this area had scattered clumps of purple loosestrife throughout (approximately 250 stems).

Finally, have you followed up with any planting of native plants or have they filled in the blank spots left when the weeds are killed? This ties in with question #4.

To date (1-2 years after treatment) only a handful natives have moved in where the dense Phragmites clones once stood. It appears that it will take sometime before natives fully repopulate the area.

Melissa Morris, Science and Stewardship Assistant, TNC-New Jersey

**IID. Case study: Control efforts in Washington State**

I've been involved with purple loosestrife (Lythrum salicaria) control for about three years on some private property here in Washington State, and the most effective method we've found is winter pulling. We have not tried any herbicides, but have done summer pulls and found winter pulls to be much more effective because: 1) the area is much wetter and plants uproot more easily and completely; 2) less biomass has to be removed and disposed of; and 3) the area can be revisited the following summer for removal of root remains that have re-sprouted. Here, the wetland freezes only sporadically during the winter, and we make a Christmas-time party out of it to get volunteers. Incidentally, we have also tried release of loosestrife-eating beetles (Galerucella pusilla, G. calmariensis), for bio-control, but although I've heard good reports about them in other areas, they didn't seem to have been able to persist after the first release on our site.

**Questions:**

**How do you uproot these monsters...just grab and pull...since all that is left in the winter are the brown dead stalks? Do you use a spade? Is there much soil disturbance with this method?**

Indeed, it's grab and pull. I've found that this is much harder in the summer, and you have to use a spade, but here the area is wet enough in

winter, and there's enough of a stalk left, that a good, careful pull gets almost all of the root-ball out. The few bits that break off you can catch the next summer when they reveal themselves by re-sprouting. The grab and pull is actually quite satisfying! And since it's so wet, it's much easier to get the excess soil off. I've done this for two winters now and will never go back to summer pulls or cuts (tried cuts too and found them much less effective).

**Are the old seed heads bagged first? Or have the dead seed heads already dispersed their seeds?**

One winter, we pulled in the same place where we had earlier cut seed heads, so they were already gone. The next year (this last one), we did not deal with the seed heads separately at all. I suppose to be most thorough, this would be a good idea, but I've found the winter pull (followed by summer touch-up) so much more effective than anything else that we didn't put extra energy into bagging seed heads, reckoning we'll be monitoring this situation for many years to come and will catch seeds if they get far enough to come up. (Also, there are nearby loosestrife areas not being controlled, and they'll be producing plenty of seed we can't control)

**Is there a lot of re-sprouting the next year? Can you actually eradicate this species after a number of years?**

In two years (July 98-now) in the area where we've been doing this method, we've pretty much gotten rid of the loosestrife, although as I said I'm sure there will be a need for ongoing monitoring of stray seed- and root-sproutings. The loosestrife in this area when we started was established and rapidly-expanding, but not yet an uninterrupted mat; now it's open water, rushes and other emergents again. This is in contrast to adjacent areas where there's been some cutting of seed heads and some no-control-at-all, where the loosestrife is continuing its relentless takeover. I plan to try another beetle release in some of these other areas, where the loosestrife mat is very dense, and continue to use the winter-pull method to reduce infested areas at the edges and stop any new invasions.

**How do you keep volunteers out in the cold water? Is there any danger of hypothermia? Do you wear wetsuits (or drysuits)?**

Yes, the cold is something to consider. We were thigh-high in water, and simply made sure there were plenty of warm dry clothes and hot cocoa waiting--but it would probably be better to use wetsuits or waders. The only other factor to consider is that the bottom can be quite mucky and my boots got stuck several times before I decided to take them off. But it was only in the low-50's when we attempted the winter-pulls . It worked well to have only a few people actually that deep in the water, and several others wearing knee-boots at the edges, catching the plants the others pulled from the deep. You can make a great game of it--the root-ball makes a weighty mass at one end and the stalk is the handle--throw it javelin-style, or twirl it and let it fly in the direction of those waiting! (As you can see, we actually had fun with this, and got the loosestrife out to boot!)

Marcy Summers, The Nature Conservancy of Washington

**IIIA. Methodology: Applications of herbicide (Michigan)**

One of our fen sites has a federally-endangered butterfly, so we have had to evaluate the impacts of herbicide and trampling. We use ACCORD, which is glyphosate and wetland approved, and half the price of RODEO. Our methods have been simplified so that volunteers can do the work without a lot of thought process.

For both loosestrife (Lythrum salicaria) and Phragmites, we use a an active mix of 5% or less (ACCORD is 41% active in concentrate and we dilute 1 part concentrate to 7 parts water). Each plant is cut and treated. Loosestrife is cut "high" (just below inflorescence) so that plant will keep growing and absorb herbicide. If cut too low, the individual "gives up" on the leader and re-sprouts from the roots. For Phragmites all stems are cut low (even if they appear dead) and treated. We use a

PVC applicator with sponge tip (design available from my MI office, contact Sara Ross, or off-of Intranet tools of the trade). The loosestrife is wiped up the stem and then the cut is also treated; only the cut is treated on the Phragmites., but more herbicide is used on Phragmites so that the strawlike structure absorbs the mix.

We also use this PVC applicator and ACCORD on woody plants, 14% active in mix (1:2) with great success. Our treatment is in patches and stepwise throughout growing season, but most intense when loosestrife is blooming (easier to locate), and in late July/August for Phragmites. To reduce trampling we try to travel through the densest patches as we treat, and have a 1 or two person team for the isolated bunches.

As for glyphosate and herps...we try to avoid dripping any mix into water, and use the wipe method since we can more carefully control the herbicide. We do not spray unless we are in solid patches, and that is on a case-by-case decision.

Jack McGowan-Stinski, Michigan Chapter, The Nature Conservancy

---

We do some control with Rodeo (glyphosate) - using what we call the Bloody Glove method, which we got from Jack McGowan-Stinski at the Michigan chapter. We are not dealing with any rare herps, but we're still looking for the best strategy for controlling the loosestrife. We do the herbiciding immediately after snapping off the flower heads, but we only work in the highest quality sites where the infestations are small.

Dave Borneman, Coordinator, Ann Arbor Department of Parks and Recreation

**The Bloody Glove technique**

We use a 5% solution of Rodeo (glyphosate; ai = 53.8%) in a spray bottle, which we add dye to. The herbicider wears chemical resistant nitrile or latex gloves on both hands covered by a fleecy, cotton glove on one of the hands. The herbicider follows behind the person removing and

bagging the flower heads. Grab the loosestrife by the hand with the cotton glove and spray the herbicide into that hand (trying not to hit non-target plants). Then wick the loosestrife up the top 1/3 of the plant. As the cotton glove becomes saturated with herbicide, you can spray less often. After herbiciding, we put the cotton gloves in a plastic container and leave the lid cracked. (If you put the lid all the way on, it can become moldy.) We can usually re-use the cotton ("bloody") gloves for quite awhile. And of course you can just rinse off the nitrile or latex gloves.

The most challenging part of the herbiciding process is not getting the herbicide all over you. Sometimes it can get tricky to tell where you have herbicided and you find yourself walking through herbicide coated loosestrife. Because of this, we require our crew to wear chemical resistant Tyvek when controlling loosestrife since it can get messy. That aside, we have found this technique to be very effective.

Kristin Condict, Technician, Ann Arbor Department of Parks and Recreation

**IIIB. Adaptive management for weed control**

I am working on a similar problem in an inland salt marsh, which has some obviously different properties from a fen. Below, see the adaptive management approach we have been using for the last 5 years, and are seriously adapting this year.

The repeated mowing had reduced the vigor of the Phragmites stand slightly but was adding lots of organic matter to the system, lots of trampling and really was not accomplishing what we needed it to. We are not managing purple loosestrife in the system, which is abundant but does not infiltrate the core salt marsh area. Beetles have been introduced in the area by USFWS, however, so we will see about that...

Bill Patterson, Field Representative, TNC - Central/Western New York Chapter

---

**Carncross Preserve Project Plan: Developing Capacity for Invasive Plant Monitoring and Control**

Project Plan
Carncross Preserve contains a globally rare, inland salt marsh community unique in its status as the highest quality example the community type in the Montezuma Wetlands complex and New York State. Distinct in species composition and ecological function from coastal salt marsh systems, inland salt marsh communities are formed by localized underlying salt deposits. Acquisition at Carncross of 100 acres of salt marsh habitat and buffer by The Nature Conservancy (TNC) in the 1980's has protected the site from salt mining, agriculture, and hydrologic alteration. However, the viability of the site remains threatened by the presence of invasive species including purple loosestrife and phragmites. Purple loosestrife, with it's low salt tolerance, represents only a mild threat to the core salt marsh area, yet the highly invasive and salt tolerant phragmites clones present at the site threaten to obliterate the salt marsh community if left unmanaged.

To address the phragmites problem at Carncross, TNC staff and volunteers began mowing the phragmites in 1995 and laid out 3 twenty-meter transects to evaluate the results. While mowing has visually reduced the vigor of phragmites, no advance or retreat of the phragmites is observed due to mowing. Monitoring of transects confirms this observation. Based on information available in The Nature Conservancy's Element Stewardship Abstracts, herbicidal control of phragmites is highly effective and can be implemented with minimal residual damage.

Management Objective:
Restoration and maintain the native inland salt marsh plant community at Carncross salt pond to 100% of potential area by eliminating (0% cover) invasive phragmites clones by spring 2003.

Monitoring Objectives:
1. Through the use of existing aerial photos and site visits, determine the extent of the potential and existing inland salt marsh community as a target for restoration.
2. Track effectiveness of herbicide applications over mud flats through the use of transects to locate the position of three representative phragmites clones.
3. In 2000 evaluate three separate methods for herbicide treatment of phragmites over native salt marsh plants while minimizing residual damage.

Note: Similar transects with photos were completed in 1995, 1996, 1997, 1998, and 1999. Data from the transect are summarized in Jim Marks' report dated November 13, 1998. Various sequences of photos from these years can be found in the Carncross files.

**1. Aerial Photos**- Aerial photos from 1999, 1989, and earlier if available to will be analyzed to map the current and potential extent of inland salt marsh community potential habitat. Location of phragmites clones will be overlaid to show extent of invasions. Identification of boundaries will occur on the ground and be related to aerial photos by GIS digitizing if available. Inland salt marsh community will be defined as the presence of a suite of inland salt marsh community indicator species such as Spartina sanisarodies, Hordeum jubatum, agropyron repens, quenapodium sp., Scirpus americanus, Scirpus meridamus, among others and is generally found between a lower mud flat boundary and an upper boundary defined by the salt intolerant purple loosestrife.

**2. Phragmites Clone Transects-**
a. Three transects A, B, and C were established in 1995 according to the appended diagram.
b. Transects consist of 10 rebar stakes 2 meters apart. Numbered 1-10 as indicated on the diagram.
c. Transects are monitored as follows.
i. Extend a tape measure the length of transect- this assists visual determination of perpendicular.
ii. Estimate the salt marsh-ward end of the phragmites clone and mark it with a string or flexible rope.
iii. Measure to the nearest decimeter, the distance (perpendicular to the base line of the transect) to the string marking the end of the clone.
iv. Measure to the nearest decimeter the distance (perpendicular to the base line of the transect) in the opposite direction (from measurements in #3) to the edge of the mud flat where the inland salt marsh community species first intersect the tape measure. Be sure to keep tape measure perpendicular to the base line.

d. Transect Photomonitoring
i. Take photos with a Kodak DC-290 digital camera at the "better" (level 2 of 4) quality setting. Zoom lens should be all the way out (widest-angle possible).
ii. Place a 3-meter tall range pole marked every 50cm. at the far end of the transect, the field of view should be centered horizontally on the pole. Vertically the field of view should focus low on the phragmites patch with only a small amount of sky showing above the 3-meter pole.
iii. One photo should be taken at each end of the 3 transects looking back down the transect.
iv. A panoramic photo is taken from the lone rebar located on the mud flat between the north basin and the south basin between transects A and B. Again the camera lens is zoomed all the way out (wide angle) and the first photo of the sequence is centered on a second lone rebar located North of the camera location. Tripod should be centered 30 degrees clockwise for each successive photo (center on 0, 30, 60, 90, 120.330). The range pole should be held at 10 meters from the lens and height of the phragmites clone at that point should be estimated on the range pole for that point and noted on the data sheet.

Notes:
Photomonitoring and transect measurements should occur in July or early August each year before any new treatments occur.
Time for transects and photomonitoring with two people = +/- 4 hours.

**3. Evaluation of Herbicide Treatments**
A variety of phragmites herbicide application techniques will be developed and evaluated depending on characteristics of the vegetation in the immediate area.
a. All phragmites clones over pure mud flats will be sprayed with glyphosate in September 2000 using a back pack sprayer. Clones will be mowed in early August to allow resprouts to attain a convenient height for herbicide application (between knee and waist high). Success will be evaluated by transects described in part 2.
b. Phragmites clones mixed with native plant communities will be controlled in 10-meter plots during year one using three experimental techniques. The most effective and time efficient techniques will be used in 2001 and 2002 to complete the restoration efforts across the entire affected area.

**i. Method 1**- In a subjectively selected location with only few widely spaced phragmites stems, use stakes to mark a 10x10-meter plot. Individually clip all phragmites stems within 10 m plot while simultaneously applying glyphosate concentrate with a Klip Kleen system. Count each stem clipped with a hand held counter. Monitoring will consist of counting phragmites stems within the plot same plot in August 2001. Photographs and visual estimates will be used to document the level of collateral damage to native plants. Photos should include close-ups of any collateral damage as well as before and after treatment diagonals showing plot corners. Logistical considerations and the amount of time to complete treatment of plot will be noted.
**ii. Method 2**- In a subjectively selected location with dense phragmites stems over native vegetation, use stakes to mark a 10x10-meter plot. Mow the entire plot in early August 2000. Broadcast spray resprouts with a backpack sprayer before phragmites senesce in 2000. Photomonitor as in method 1, stem counts will not apply.
**iii. Method 3**- In a subjectively selected location with dense phragmites stems over native vegetation, use stakes to mark a 10x10-meter plot. Mow the entire plot in early August 2000. Use a wet glove or cloth herbicide application method to treat all individual phragmites stems and monitor as in method 2.

During summer 2001, the most efficient (least time and least residual kill compromise) will be used to eradicate phragmites from the potential inland salt community area.

**IV. Suggested references**

**Melissa Morris (New Jersey Chapter Office) suggests these references on the effect (or lack thereof) of glyphosate or other herbicides on herptiles, specifically turtles:**

Brooks, J.J., A. S. Johnson, and K.V. Miller. 1993. Effects of chemical site preparation on wildlife habitat and plant species diversity in the Georgia Sandhills. Pp. 605-612 in J.C.
Brissette, ed. Proceedings of the Seventh Biennial Southern Silviculture Research Conference. United States Department of Agriculture, Forest Service, General Technical Report SO-93.
Cole, E.C., W.C. McComb, M. Newton, C.L. Chambers, and J.P. Leeming. 1995. Responses of small mammal and amphibian capture rates to clear cutting, burning and glyphosate application in the Oregon range. Pp. 155-157 in R.E. Gaskin and J.A. Zabkiewicz, compilers. Second International Conference on Forest Vegetation Management, Rotorua, New Zealand. New Zealand Forest Research Institute Bulletin Number 192.

Howell, D., Miller, K., Bush, P. and J. Taylor. 1996. Herbicides and Wildlife Habitat (1954-1996): An Annotated Bibliography on the Effects of Herbicides on Wildlife Habitat and Their Uses in Habitat Management. USDA Forest Service. Technical Publication R8-TP 13 (revised).

**Further reference suggestions:**
I don't have experience using herbicides in fens, but I have seen some literature on the residence time of some of the herbicides and the residues they break down to in the soil. Some can last a fairly long time, despite the manufacturer's claims. There is also some evidence that plants translocate the stuff down to their roots, making treating cut-stems a problem.

My recollection was that most of the fens were in pretty good shape with isolated areas of invasives. I think it might be useful, if you haven't done it already, to monitor bog turtle movement and nesting using radio tracking and other means. Also, I think your idea of treating in stages, while not optimal, may be a good compromise.

I would think if the volunteers were careful, trampling shouldn't be a problem if you have some idea where nesting is occurring. Nesting at one site, as I recall, was in a rather small area, though maybe your studies have shown otherwise since then. So, if that's true at your site, you could treat other areas outside of the nesting areas. Anyway, check out the literature on residence time of these chemicals.

Michael S. Batcher, M.S., A.I.C.P., Consulting Ecologist and Environmental Planner

**V. Contact Information**

Michael S. Batcher, M.S., A.I.C.P.
Consulting Ecologist and Environmental Planner
1907 Buskirk-West Hoosick Rd.
Buskirk, NY 12028
Ph: (518) 686-5868; FAX: (518) 686-1802;
E-mail: mbatcher@netheaven.com

Dave Borneman, Coordinator
Natural Area Preservation Division
Ann Arbor Department of Parks and Recreation
1831 Traver Rd. Ann Arbor, MI 48105
Ph: (734) 994-4834; FAX: (734) 997-1072;
E-mail: dborneman@ci.ann-arbor.mi.us

Kristin Condict, Technician
Natural Area Preservation Division
Ann Arbor Department of Parks and Recreation
Ph: (734) 996-3266; FAX: (734) 997-1072

Betsy Lyman, Assistant Director of Science & Stewardship
The Nature Conservancy - Pennsylvania Field Office Lee Park,
1100 East Hector St., Suite 470
Conshohocken, PA 19428
Ph: (610) 834-1323 ext. 103; FAX: (610) 834-6533;
E-mail: blyman@tnc.org

Jack McGowan-Stinski
The Nature Conservancy - Michigan Chapter
2840 East Grand River Ave., Suite 5
East Lansing, MI 48823
Ph: (517) 332-1741; FAX: (517) 332-8382;
E-mail: jmcgowan-st@tnc.org

Melissa Morris, Science and Stewardship Assistant
The Nature Conservancy - New Jersey Chapter Office
Ph: (908) 879-7262; FAX: (908) 879-2172;
E-mail: mmorris@tnc.org

Mike Norris, Restoration Specialist
Southern Lake Michigan Rim Project
Ph: (219) 473-4312

Chip O'Leary, Restoration Coordinator
The Nature Conservancy - Indiana Chapter
2400 New York Ave.
Whiting, IN 46394
Ph: (219) 473-4312;
E-mail: c'oleary@tnc.org or olearych@aol.com

Bill Patterson, Field Representative
The Nature Conservancy - Central/Western New York Chapter
Ph: (716) 546-8030 ext. 24

Marcy Summers
The Nature Conservancy of Washington
217 Pine St., Suite 1100
Seattle, WA 98101
Ph: (206) 343-4344 ext. 323; FAX: (206) 343-5608;
E-mail: msummers@tnc.org

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




# Japanese knotweed and Giant knotweed
# *Polygonum cuspidatum* Siebold and Zucc. and *Polygonum sachalinense* F.W. Schmidt ex. Maxim.

**SOURCE**: The following information on this species is taken, in part, from the Delaware River Invasive Plant Partnership, Invasive Species Fact Sheets, http://www.paflora.org/DRIPP.html. Modifications include additional pictures from www.invasives.org and http://invasives.eeb.uconn.edu/ipane/, as well as additional resource information. Check the website links for future updates. The last update for this fact sheet is April, 2002.

**NATIVE RANGE:** Japan
**DESCRIPTION:** Japanese knotweed and giant knotweed are herbaceous perennials that form large colonies of erect stems that can reach 9 feet in height. They spread by vigorous rhizomes (horizontal stems that grow just below the soil surface). Japanese knotweed and giant knotweed are very similar in appearance and are known to hybridize. The best character for separating them is the shape of the leaf base, those of Japanese knotweed are truncate (squared-off) at the bottom, while those of giant knotweed are heart-shaped.

**Height** - Individual stems are 3-9 feet tall depending on the vigor of the colony.
**Stem** - The hollow, bamboo-like stems are erect and unbranched or with a few branches toward the tip. Despite their size, knotweed stems are annual; they die back to the rhizome at the end of the growing season. New shoots emerge in April and grow rapidly; early in the season they can grow 3-4 inches per day.
**Leaves** - Leaves are alternate on the stem, simple, 4-6 inches long and almost as wide, and dark green. Japanese knotweed leaves are abruptly squared-off (truncate) at the base; those of giant knotweed have a heart-shaped base. Both narrow to a pointed tip.
**Flowers** - Both Japanese knotweed and giant knotweed have numerous small, greenish-white lowers that are produced in late summer. Japanese knotweed bears only male or female flowers on a given plant. Giant knotweed blooms have both male and female parts in the same flower. However, appearances can be difficult to interpret as both the male and female flowers of Japanese knotweed have vestigial organs of the other sex present.
**Fruit and seed** - The seed (technically a fruit called an achene) of both knotweeds is shiny black, 3-angled, and about 1/6 inch long. It is enclosed in a winged calyx that contributes to its buoyancy. The seeds have no dormancy requirement and germinate readily.
**Roots** - Roots are present along the rhizome and can extend quite deeply into the soil making knotweed effective in preventing erosion.








Photos: Leslie J. Mehrhoff, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/ and Jil W. Swearingen, USDI National Park Service, www.forestryimages.org.

Counterclockwise: Fruits (Jil Swearingen), Flowering plants, Comparison of leaves of *P. sachalinense* (right of each pair) and *P. cuspidatum*, Stand of young plants, cross-section of hollow stem.





**DISTRIBUTION AND HABITAT:** Japanese knotweed is native to Japan; giant knotweed comes from Sakhalin Island in northern Japan. They were introduced into North America for ornamental use in the late 1800s. Japanese knotweed is now widely naturalized in Europe and North America. In the east it extends from Newfoundland to North Carolina. It is also widespread in the Midwest and in coastal areas of the Pacific Northwest. It is most commonly found lining the banks of creeks and rivers where it often forms an impenetrable wall of stems; it also occurs in wetlands, waste ground, and along roads and railroads. In Pennsylvania knotweed has also been extensively planted at strip mine reclamation sites.

**EFFECTS OF INVASION:** Dense stands of knotweed exclude other plant species leading to very limited biological diversity in infested sites.



Photo: Leslie J. Mehrhoff, University of Connecticut, IPANE, http://invasives.eeb.uconn.edu/ipane/

**REPRODUCTION AND METHODS OF DISPERSAL**: Japanese knotweed and giant knotweed both spread vegetatively by the growth and fragmentation of rhizomes. Even a 1-2 inch-long piece of rhizome dislodged by flooding can initiate a new colony when it is deposited downstream. Knotweed also grows from seeds, which are produced in large numbers and dispersed by wind and water. Seed viability is high, and seed bank densities have been measured at 220-1758 seeds per square meter. Highest germination rates occur on exposed mineral soil.

**NATIVE ALTERNATIVES FOR REVEGETATION OF STREAM BANKS:** The following species are suggested for establishing native plant cover after knotweed has been removed: shrubs - winterberry holly (*Ilex verticillata*), spicebush (*Lindera benzoin*), buttonbush (*Cephalanthus* occidentalis), silky willow (Salix sericea), pussy willow (Salix discolor), American elderberry (Sambucus canadensis), alder (Alnus serrulata and *A. incana* ssp. *rugosa*); herbaceous species- riverbank rye (*Elymus riparius*), wild-rye (*Elymus villosus*), big bluestem (*Andropogon gerardii*), switch grass (*Panicum virgatum*), wingstem (*Verbesina alternifolia*), joe-pye-weed (*Eupatorium fistulosum* and *E. maculatum*), boneset (*Eupatorium perfoliatum*).

**AUTHORS:**
Ann F. Rhoads and Timothy A. Block, Morris Arboretum, University of Pennsylvania.

**For more information on Japanese and Giant knotweed, please contact:**
Cornell University, Biological Control of Weeds--Garlic Mustard, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Delaware River Invasive Plant Partnership, http://www.paflora.org/DRIPP.html
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Ohio Department of Natural Resources, Division of Natural Areas and Preserves, http://www.dnr.state.oh.us/dnap/invasive/10japknotweed.htm
Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild Fact Sheets, http://www.nps.gov/plants/alien/fact/pocu1.htm.
Plant Invaders of Mid-Atlantic Natural Areas, http://www.nps.gov/plants/alien/pubs/midatlantic/
Southeast Exotic Pest Plant Council; Invasive Plant Manual, http://www.se-eppc.org/manual/
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program Fact Sheet, Japanese knotweed, http://www.dcr.virginia.gov/dnh/invlist.htm

Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm

**REFERENCES:**

McCormick, L. H. and T. W. Bowersox. 1998. Eradication and control of Japanese knotweed at the Staple Bend Unit, Allegheny Portage Railroad National Historic Site. Penn State School of Forest Resources, University Park, PA.
Niewinski, A. T., T. W. Bowersox, and L. H. McCormick. 1999. Reproductive ecology of giant (Polygonum sachalinensis) and Japanese (Polygonum cuspidatum) knotweed. National Park Service Technical Report NPS/PHSO/NRTR-00/079. University Park, PA.
Reeder, Kathleen Kodish and Brian Eick. 2001. Northeast parks' regional strategy to control knotweed. Park Science 21: 33-35.
Rhoads, Ann Fowler and Timothy A. Block. 2000. The Plants of Pennsylvania: An Illustrated Manual. University ofPennsylvania Press, Philadelphia, PA.
Rhoads, Ann Fowler and William McKinley Klein. 1993. The Vascular Flora of Pennsylvania: Annotated Checklistand Atlas. American Philosophical Society, Philadelphia, PA.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Japanese knotweed and Giant knotweed
# *Polygonum cuspidatum* Siebold and Zucc. and
# *Polygonum sachalinense* F.W. Schmidt ex. Maxim.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Japanese knotweed - http://tncweeds.ucdavis.edu/esadocs/polycusp.html - Author: Leslie Sieger - 1991.**

Current control methods (mechanical, herbicidal) require continued treatment to prevent reestablishment of Polygonum cuspidatum. It may be feasible to reintroduce competitors as an alternative to continued treatment. There is a need for more research on whether native species might serve effectively as competitors and methods of reintroduction. Only very preliminary work has been done towards developing a biological control for Polygonum cuspidatum and much research remains to be done (see below).

In areas where Polygonum cuspidatum has not yet become established, the focus of management should be to prevent establishment by monitoring areas for introductions of Polygonum cuspidatum and eradicating newly established stands before they can become established.

## MANUAL AND MECHANICAL CONTROL:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Japanese knotweed and Giant knotweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April, 2002.**

Repeated cutting of the stems reduces vigor and with persistence might be sufficient to control small, isolated populations. Attempts to dig out the plants are doomed to fail because of the ability of even small segments of rhizome to resprout.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #10 - Japanese knotweed - http://www.dnr.state.oh.us/dnap/invasive/10japknotweed.htm - January 15, 2003**

Large colonies of this species are extremely difficult to dig up due to their high rhizome densities. Digging of large colonies is not recommended as it is very labor intensive and unlikely that all below ground material can be removed. Small patches may be dug, however care should be used in removing plant material as improper disposal can spread the species further. Repetitive cutting or mowing within a single growing season to deplete stored reserves and remove photosynthetic tissue has been effective. Eradication of the rhizome system is necessary for control of this aggressive invasive species.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Japanese knotweed- http://www.nps.gov/plants/alien/fact/pocu1.htm - Author: Tom Remaley - May 5, 2004.**

Grubbing is effective for small initial populations or environmentally sensitive areas where herbicides cannot be used. Using a pulaski or similar digging tool, remove the entire plant including all roots and runners. Juvenile plants can be hand pulled depending on soil conditions and root development. Any portions of the root system not removed will potentially resprout. All plant parts (including mature fruit) should be bagged and

disposed of in a trash dumpster to prevent reestablishment.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Japanese knotweed - http://www.se-eppc.org/manual/knotweed.html - 2003**

Grubbing: This method is appropriate for small initial populations or environmentally sensitive areas where herbicides cannot be used. Using a pulaski or similar digging tool, remove the entire plant including all roots and runners. Juvenile plants can be hand-pulled depending on soil conditions and root development. Any portions of the root system not removed will potentially resprout. All plant parts, including mature fruit, should be bagged and disposed of in a trash dumpster to prevent reestablishment.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Japanese knotweed - http://tncweeds.ucdavis.edu/esadocs/polycusp.html - Author: Leslie Sieger - 1991.**

Manual control consists of digging out the rhizomes or cutting the stalks. Digging is extremely labor intensive and tends to spread the rhizome fragments and promote disturbance and is not recommended (Palmer, 1990). Cutting, on the other hand, may be quite effective in eliminating Polygonum cuspidatum. It has been observed that Polygonum cuspidatum does not establish where grazing pressure is high (Beerling, 1990; Palmer, 1990). In a review of control methods, Palmer (1990) noted that eradication is not complete with cutting alone, but has been nearly achieved in some cases and should be feasible with persistence. A number of authors claim that cutting is ineffective (Pauly, 1986; Pridham and Bing, 1975; Orchowski, 1991). These conclusions are based on the observation that shoots are regenerated following cutting. However, a greenhouse study found that cutting stems results in a significant reduction of rhizomatous reserves. The same study also found that cutting was equally effective at any time during the growing season prior to the beginng of senescence (Seiger and Merchant, 1990). A study of the effects of repeated cutting showed that at least three cuts are needed in a growing season to offset rhizome production (Seiger, unpublished data). Manual control can be labour intensive, but where populations are small and isolated, may be the best option. No research has been done to test the effectiveness of burning. It may act similarly to cutting by removing above ground material.

Shading, particularly in conjunction with cutting, may be another useful means of control on smaller stands. Studies showing that P. cuspidatum requires high light environments suggest that covering stands with black plastic or shade cloth may reduce growth. Pridham and Bing (1975) state that applying several layers of black polyethylene film covered by asphalt, blocks or stones to a leveled soil surface may provide some control. However, they also note that P. cuspidatum is able to emerge through asphalt. If shade cloth (or plastic) is to be applied without cutting, then, to prevent P. cuspidatum from emerging through the covering, shade cloth should be placed over shoots after the plants have reached their full height or placed well above newly emerging shoots, or raised as plants grow.

## BIOCONTROL:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Japanese knotweed and Giant knotweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April, 2002.**

No biological control options are currently available.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #10 - Japanese knotweed - http://www.dnr.state.oh.us/dnap/invasive/10japknotweed.htm - January 15, 2003**

There are currently no biological controls available for Japanese knotweed.

**USDA Forest Service - Biological Control of Invasive Plants in the Eastern United States, http://www.invasive.org/eastern/biocontrol/12Knotweed.html - Publication FHTET-2002-04 - Authors: R.Van Driesche, et al. - 2002,**

The biological control of this weed has not yet been attempted, although projects in the United Kingdom and United States are being considered.

## CHEMICAL CONTROL*:

**Delaware River Invasive Plant Partnership - Invasive Plant Fact Sheets - Japanese knotweed and Giant knotweed - http://www.paflora.org/DRIPP.html - Authors: Ann F. Rhoads and Timothy A. Block - April, 2002.**

Research conducted at Penn State for the National Park service resulted in a recommendation of a foliar spray of glyphosate plus sticker-spreader applied in early June and winged calyx which encloses the fruit (achene) of P. cuspidatum again in late August of the same year at the rate of 4 lbs active ingredient per acre. A third application may be needed the following spring if significant regrowth occurs. Rapid establishment of alternative plant cover is an important aspect of control as knotweed seedlings do not compete well with other vegetation.

The British Nature Conservancy Council recommends cutting in late spring or summer followed by an application of glyphosate in the fall. At least two additional applications will be needed to control the regrowth.

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #10 - Japanese knotweed - http://www.dnr.state.oh.us/dnap/invasive/10japknotweed.htm - January 15, 2003**

Herbicide has been generally effective at controlling this species. Repetitive cutting of stems with spot application of Roundup®, Accord® or Glypro® to the stumps, and foliar spraying in large populations has been reported to be successful.

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Japanese knotweed-**

**http://www.nps.gov/plants/alien/fact/pocu1.htm - Author: Tom Remaley - May 5, 2004.**

Cut stem treatment: Use this method in areas where plants are established within or around non-target plants or where vines have grown into the canopy. This treatment remains effective at low temperatures as long as the ground is not frozen. Cut the stem about 2 inches above ground level. Immediately apply a 25% solution of glyphosate (e.g., Roundup, or use Rodeo if applying in or near wetland areas) or triclopyr (e.g., Garlon) and water to the cross-section of the stem. A subsequent foliar application of glyphosate may be require to control new seedlings and resprouts.

Foliar spray method: Use this method to control large populations. It may be necessary to precede foliar applications with stump treatments to reduce the risk of damaging non-target species. Apply a 2% solution of glyphosate or triclopyr and water to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. A 0.5% non-ionic surfactant is recommended in order to penetrate the leaf cuticle, and ambient air temperature should be above 65 °F.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Japanese knotweed - http://www.se-eppc.org/manual/knotweed.html - 2003**

Foliar Spray Method: This method should be considered for large thickets of bush honeysuckle where risk to non-target species is minimal. Air temperature should be above 65Â°F to ensure absorption of herbicides.

> Glyphosate: Apply a 2% solution of glyphosate and water plus a 0.5% non-ionic surfactant to thoroughly wet all leaves. Use a low pressure and coarse spray pattern to reduce spray drift damage to non-target species. Glyphosate is a non-selective systemic herbicide that may kill non-target partially-sprayed plants.
> Triclopyr: Apply a 2% solution of triclopyr and water plus a 0.5% non-ionic surfactant to thoroughly wet all leaves. Use a low pressure and coarse spray pattern to reduce spray drift damage to non-target species. Triclopyr is a selective herbicide for broadleaf species. In areas where desirable grasses are growing under or around bush honeysuckle, triclopyr can be used without non-target damage.

Cut Stump Method: This control method should be considered when treating individual bushes or where the presence of desirable species precludes foliar application. This treatment is effective as long as the ground is not frozen.

> Glyphosate: Horizontally cut bush honeysuckle stems at or near ground level. Immediately apply a 25% solution of glyphosate and water to the cut stump, covering the outer 20% of the stump.
> Triclopyr: Horizontally cut bush honeysuckle stems at or near ground level. Immediately apply a 25% solution of triclopyr and water to the cut stump, covering the outer 20% of the stump.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Japanese knotweed - http://tncweeds.ucdavis.edu/esadocs/polycusp.html - Author: Leslie Sieger - 1991.**

A number of biocidal chemicals have been found to be effective against Polygonum cuspidatum. Most of these are undesirable for use in conservation areas because they are nonselective, may be persistent in the soil and/or are not safe for use near water. One frequently used way to minimize the effects of non-selective herbicides on non-target species is to paint herbicides directly onto the target plants (Broaddus, pers. comm.). In the case of P. cuspidatum, this would probably require prior cutting for easier access if herbicides are to be applied after the plants have reached their full height. Herbicides appear to be more effective when combined with cutting (Scott and Mars, 1984; Orchowski, 1991).

Glyphosate [N-(phophonomethyl)glycine] has been found to be very effective against Polygonum cuspidatum (Ahrens, 1975; Beerling, 1990; Pauly, 1986). Glyphosate is a nonselective systemic herbicide with a short residual life (Ahrens, 1975; Lynn, Rogers and Graham,1979). Application is more effective in the fall when leaves are translocating to rhizomes (Lynn, et al, 1979). The British Nature Conservancy Council (1989) recommends applying 2.0 kg/ha in August with a prior cut in late spring or early summer. Glyphosate is available from Monsanto under the trade names RoundupTM and RodeoTM. Only Rodeo has been approved for use near water (Bender, 1988). Glyphosate has been used with limited success on some nature reserves in the U.K. (Palmer, 1990). Repeated applications over several years may be necessary (Beerling, 1990; Palmer, 1990; Pauly, 1986).

The Nature Conservancy Council (1989) also recommends picloram to be applied at a rate of 2.6 kg/ha in the spring. Picloram is a selective herbicide which is persistent in the soil. It must not be used near water, thus excluding its use in many of the areas where Polygonum cuspidatum is a problem (Gritten, 1990; Scott and Mars, 1984). Dicamba (3,6-dichloro-o-anisic acid) has also been found to be effective against Polygonum cuspidatum, but is persistent in the soil and nonselective (Pridham and Bing, 1975). A number of other herbicides have been tested against Polygonum cuspidatum, both alone and in combination with other herbicides (Orchowski, 1991; Scott and Mars, 1984). Herbicide may have to be used on stands that have been allowed to attain a large size. However, their use is not recommended in nature reserves because of their undesirable effects on other biota and the need for repeated applications to maintain control of Polygonum cuspidatum.

Regardless of whether control is manual or chemical, as long as some rhizomes remain in the soil Polygonum cuspidatum returns once management is relaxed (Beerling, 1990; Nature Conservancy Council, 1989; Palmer, 1990). It has been suggested that the reintroduction of effective competition might be possible (Eaton, 1986).

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Invasive Species Initiative - Controlling knotweed in the PNW - http://tncweeds.ucdavis.edu/esadocs/polycusp.html - Jonathon Soll.**

A **great reference** paper by Jonathan Soll of TNC-OR, who tested various control methods for Japanese and Giant knotweed. This document is too large to include in this Management and Control document.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Japanese stiltgrass, Nepalese browntop
## Microstegium vimineum (Trin.) Camus

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org and http://tncweeds.ucdavis.edu/photos.html) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is May, 2004.

**NATIVE RANGE**: Europe

**DESCRIPTION**: Lesser celandine, also known as fig buttercup, is an herbaceous, perennial plant in the buttercup family (Ranunculaceae).



Photo: © Mandy Tu/The Nature Conservancy

Plants have a basal rosette of dark green, shiny, stalked leaves that are kidney- to heart-shaped.



Photo: Leslie J. Mehrhoff, Univ. of Connecticut, www.invasive.org.

The flowers open in March and April, have eight glossy, butter-yellow petals, and are borne singly on delicate stalks that rise above the leaves. Pale-colored bulblets are produced along the stems of the above-ground portions of the plant, but are not apparent until late in the flowering period. When in bloom, large infestations of lesser celandine appear as a green carpet with yellow dots, spreading across the forest floor. There are many varieties of lesser celandine including a double-flowered form with many crowded petals and dark green leaves mottled with silvery markings.

NOTE: Lesser celandine closely resembles marsh marigold (Caltha palustris), a native wetland plant that occurs in the eastern United States.

Marsh marigold is a robust plant with glossy, rounded or kidney-shaped leaves and flowers on stalks that are 8 in (20.3 cm) or more in height and consist of five to nine deep yellow "petals" (actually sepals). Marsh marigold does not produce tubers or bulblets, nor does it form a continuous carpet of growth. Extreme care should be taken to correctly identify lesser celandine before undertaking any control measures to avoid impacts to this plant.

**ECOLOGICAL THREAT**: Lesser celandine is an exotic spring ephemeral and a vigorous growing groundcover that forms large, dense patches on the forest floor, displacing and preventing native plants from co-occurring.



Photo: Leslie J. Mehrhoff, Univ. of Connecticut, www.invasive.org.

The ecological impact of lesser celandine is primarily on the native spring-flowering plant community and the various wildlife species associated with them. Spring ephemerals complete the reproductive part of their life cycle and most of their above-ground development before woody plants leaf out and shade the forest floor. Native spring ephemerals include bloodroot, common and cut-leaved toothwort, Dutchman's breeches, harbinger-of-spring, squirrel-corn, trout lily, Virginia bluebells, and many others. Because lesser celandine emerges well in advance of the native species, it can establish and overtake areas rapidly.

**DISTRIBUTION IN THE UNITED STATES**: Lesser celandine is currently found in nineteen states in the Northeast and Pacific Northwest (USDA PLANTS map). It is reported to be invasive in nine states (Connecticut, Delaware, Maryland, New Jersey, Oregon, Pennsylvania, Virginia, Wisconsin, West Virginia), and the District of Columbia (WeedUS Database).

**HABITAT IN THE UNITED STATES**: Lesser celandine occurs in moist forested floodplains and in some drier upland areas, and seems to prefer sandy soils.

**BACKGROUND**: Lesser celandine was introduced to the United States as an ornamental plant. It is still available commercially in the U.S., along with many colorful varieties. All varieties of lesser celandine should be assumed to be invasive.

**BIOLOGY & SPREAD**: Lesser celandine is an exotic perennial plant and spring ephemeral that spends much of the year (summer through early winter) underground as thickened, fingerlike tubers or underground stems. During the winter, leaves begin to emerge and photosynthesize in preparation for flowering. Flowering usually occurs from late winter through mid-spring (March through May), depending on conditions. Afterwards, the above-ground portions die back. Lesser celandine spreads primarily by vegetative means through abundant tubers and bulblets, each of which is ready to become a new plant once separated from the parent plant. The tubers of lesser celandine are prolific and may be unearthed and scattered by the digging activities of some animals, including well-meaning weed pullers, and transported during flood events.

**For more information on lesser celandine, please contact:**
Sue Salmons, Natural Resources Manager, National Park Service, Rock Creek Park, Washington, DC (sue_salmons@nps.gov)

Jil Swearingen, Integrated Pest Management Coordinator, National Capital Region, Center for Urban Ecology, Washington, DC (jil_swearingen@nps.gov)
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov

**SUGGESTED ALTERNATIVE PLANTS**: Many lovely, perennial, spring-flowering plants are available as non-invasive alternatives to lesser celandine. Some examples of plants native the eastern U.S. include wild ginger (Asarum canadense), Dutchman's breeches (Dicentra cucullaria), squirrel-corn (Dicentra canadensis), cutleaf toothwort (Cardamine concatenata), twinleaf (Jeffersonia diphylla), and bloodroot (Sanguinaria canadensis). Contact your local native plant society for additional suggestions and assistance. The Plant Conservation Alliance provides links to many groups at (http://www.nps.gov/plants).

**AUTHOR**: Jil M. Swearingen, National Park Service, National Capital Region, Center for Urban Ecology, Washington, DC.

**REVIEWERS**: Carole Bergmann, Sally Gagne, Larry Morse, Ann Rhoads, Steve Young.

**REFERENCES**: Bailey, L.H. and E.Z. Bailey. 1977. Hortus Third: A Concise Dictionary of Plants Cultivated in the United States and Canada, MacMillan Publishing Co., Inc., New York.
Fernald, M. L. 1970. Gray's Manual of Botany, Eighth edition. D. Van Nostrand Company, New York, NY. p. 648.

Peterson, R.T. and M. McKenny. 1968. A field guide to wildflowers Northeastern and North-central America: Houghton-Mifflin Co., Boston, MA. 420 pp.
Salmons, S. 2003. Presentation to Mid-Atlantic Exotic Pest Plant Council, University of Pennsylvania, Philadelphia, PA.
Strasbaugh, P. D. and E. L. Core. Flora of West Virginia, Second Edition. Seneca Books, Inc. Grantsville, WV. Swearingen, J. 2004. WeedUS: Database of Invasive Plants of Natural Areas in the U.S. (in progress). http://www.nps.gov/plants/alien.

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Lesser Celandine
# *Ranunculus ficaria* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resources:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Lesser Celandine - http://www.nps.gov/plants/alien/fact/rafi1.htm - Author: Jil M. Swearingen - December 3, 2001**

## GENERAL MANAGEMENT CONSIDERATIONS:

Lesser celandine is very difficult to control but it can be managed with persistence over time using methods that are site appropriate. While manual methods are possible for some (small) infestations, the use of systemic herbicide kills the entire plant tip to root and minimizes soil disturbance.

## MANUAL AND MECHANICAL CONTROL:

Manual: For small infestations, lesser celandine may be pulled up by hand or dug up using a hand trowel or shovel. It is very important to remove all bulblets and tubers.

Mechanical: If mechanical removal is to continue after dieback of the plants, individual plants or clumps will need to be marked with some sort of stakes or flagging because it will be impossible to relocate the plants otherwise. When conducting mechanical removal, care should be taken to minimize soil disturbance as much as possible. For this reason, mechanical control may be inappropriate for large infestations in high quality natural areas.

## BIOCONTROL:

No biological control agents are currently available for lesser celandine

## CHEMICAL CONTROL:

The window of opportunity for controlling lesser celandine is very short, due to its life cycle. In order to have the greatest negative impact to celandine and the least impact to desirable native wildflower species, herbicide should be applied in late winter-early spring (March through May). Apply a 1.5% rate of a 39 to 41% glyphosate isopropylamine salt (e.g., Rodeo for wetland areas) mixed with water and a non-ionic surfactant to foliage, avoiding application to anything but the celandine. Glyphosate is systemic; that is, the active ingredient is absorbed by the plant and translocated to the roots, eventually killing the entire plant. The full effect on the plant may take 1-2 weeks. Applications can be made during the winter season as long as the temperature is above about 50 degrees Fahrenheit, and no rain is anticipated within 12 hours. Because glyphosate is non-specific, spray should be controlled such that it touches only lesser celandine and does not drift onto desirable plants. To minimize impacts to sensitive-skinned frogs and salamanders, some experts recommend applying herbicide in March and then switching to manual methods.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Narrow-leaved and Hybrid Cattail
## *Typha angustifolia* and *T. x glauca*

**SOURCE**: The following information on this species is taken, in part, from the Ohio Department of Natural Resources, Division of Natural Areas and Preserves, Invasive Plants of Ohio Fact Sheets website at http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is January 8, 2003.

**DESCRIPTION:** Narrow-leaved cattail is a non-native, invasive plant that hybridizes with the native broad-leaved cattail (*T. latifolia*) to produce the invasive *T. xglauca*. All three aquatic perennials may grow up to a height of 10 feet and produce a velvety brown spike of flowers. The flower head of the hybrid and the narrow-leaved cattail have a gap of 1-4 inches between the male and female flowers, while the native species has both flower types next to each other. The leaves of cattail originate from the base and spread outward. The narrow-leaved and hybrid cattails have leaves that are 1/4 - 3/4 inch across; the native cattail's leaves are wider at 1/2 -1 inch. A starchy rhizome forms beneath each plant.



Narrow-leaved (Left) and Broad-leaved (Right) Cattail Division Photo

Photo: Ohio Department of Natural Resources, Division of Natural Areas and Preserves, http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm



Photo of T. angustifolia: © Mandy Tu/ The Nature Conservancy

**HABITAT**: Stands of cattail can be found in a wide variety of wetland habitats, including marshes, lakeshores, river backwaters and roadside ditches. This prolific plant can grow in disturbed areas, as well as brackish, and polluted waters of depths nearing 3 feet.

**DISTRIBUTION:** Narrow-leaved cattails are believed to have been introduced to the Atlantic seaboard from the dry ballast of European ships. This plant has since spread westward and occurs throughout much of the United States. The hybrid cattail is concentrated in the northeast, but may occur wherever both the native and the narrow-leaved species are present. All three taxa are found throughout Ohio.

**PROBLEM:** Narrow-leaved and hybrid cattail will out-compete native plants in wetland systems. These plants establish dense monocultures that enable them to shade out native vegetation. They are also thought to be allelopathic, producing chemicals which discourage growth of other plant species. Cattails reproduce both vegetatively by rhizomes and sexually through massive amounts of seed.

Photo: © Mandy Tu/ The Nature Conservancy



**For more information on non-native cattails, please contact:**

Grace, J.B. and J.S. Harrison. 1986. The Biology of Canadian Weeds: Typha latifolia L., T. angustifolia L. and T. xglauca Godr. Canadian Journal of Plant Science 66: 361-379.
Hoffman, R. and K. Kearns, eds. 1997. Wisconsin Manual of Control Recommendations for Ecologically Invasive Plants. Bureau of Endangered Resources, Wisconsin Department of Natural Resources.
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
Motivans, K. and S. Apfelbaum. 1987. Element Stewardship Abstract for Typha spp.
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Ohio Department of Natural Resources, Division of Natural Areas and Preserves, http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm
Wisconsin Department of Natural Resources, Non-native plants fact sheets, Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm

**Management and Control Information**

Return to List




## Species Management and Control Information

# Narrow-leaved and Hybrid Cattail
# *Typha angustifolia* and *T. x glauca*

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

The acreage of cattail-dominated wetlands in the United States has increased drastically since the early twentieth century due to changes in hydrology and land use. The optimal control technique for a given site will depend on the hydrologic state of the site, the size of the area to be managed, and if the manager is able to manipulate water levels.

## MANUAL AND MECHANICAL CONTROL:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Manipulation of water levels can kill cattails by inhibiting airflow from the cattail shoots to the roots. Removing the dead leaves and submerging the shoots in early spring will eliminate gas diffusion and "suffocate" the plant. In situations where water level manipulations are either not feasible or appropriate, pulling, cutting and bulldozing treatments have been used with some success. In the case of bulldozing, the benefits in effective removal may not outweigh the costs of disturbing the wetland.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Typha spp. - http://tncweeds.ucdavis.edu/esadocs/typh_spp.html - Author: K. Motivans and S. Apfelbaum - October 27, 1987.**

WATER LEVEL MODIFICATION:
High water conditions in a cattail stand can affect the growth of seedlings, can break off mature stalks, or can be followed by the immigration of muskrats which eat the cattail (Zimmerman pers. comm.). The effect of flooding does not always have negative impacts on cattails -- plants have been known to float up and continue growing until water returns to a previous lower levels.

As with any control measure, temporary conditions, such as flooding, do not prevent later seed establishment. Cattail seeds can arrive from a great distance, and it doesn't take but a few seeds to germinate and rapidly produce clones as adults. The cost of management actions should be considered when dealing with unknown response variables.

Low water conditions, maintained by draining a wetland, significantly effects the overall community (Mallik and Wein 1985). Harris and Marshall (1963) concluded that draining techniques have possible detrimental effects because the plant composition of a wetland can be radically changed. Draining alone can cause a significant increase in Typha cover under some conditions (Mallik and Wein 1985). However, to inhibit Typha growth, a wetland can be drained and then burned during the summer. If there is no reserve of water over winter cattails will not survive the following spring, according to Zimmerman (pers. comm.) but there have been no controlled experiments to show this.

Two years of 65 cm (26 in) deep flooding was required before established cattail began to die and open water conditions were created at

Sinnissippi Marsh. Cattail initially survived flooding from 1973-1977 and became the dominant emergent plant. A light green color, noticeably narrower leaves, and absence of fruiting heads indicated stress in 1976. Cattail stem densities declined 57 percent with all emergent plants dead in 1977. Horicon Marsh, flooded to a depth of 40 cm (16 in), showed declines in emergent and aquatic plants. Cattail required two years before it declined (Wisconsin DNR 1969 and 1971).

Mature T. latifolia and seedlings less than one year old are killed by water depths of 63.5 cm (25 in) and 45 cm (18 in) or more, respectively. Narrow-leaved cattail was unaffected by this degree of flooding. Narrow-leaved cattail establishment was prevented when water levels were maintained at 1.2 m (47 in) or deeper (Steenis et al. 1958). Dryer conditions allowed more clones of T. angustifolia to be spread (McMillan 1959).

Because cattails can transpire significant quantities of water (2-3m of water/acre/year) (Fletcher and Elmendor 1955, Zohary 1962), their establishment may serve to exacerbate water level instability and further contribute to disruptive influences supporting increased cattail. Flooding must account for evapotranspirational losses of water to maintain a level effective in cattail control.

PHYSICAL CONTROL:
Hand or mechanical cutting of cattails followed by submergence of all cattail stems results in high control. Up to 100 percent cattail control was measured two growing seasons after treatment. No visible cattail regrowth occurred in one year and cattail rhizomes were dead. The highest cattail control of any method tested was achieved by two clippings followed by stem submergence to at least 7.5 cm (3 in) (Nelson and Dietz 1966). Control was best if plants were cut in late summer or early fall.

In Iowa (Weller 1975), cutting cattail and reflooding with at least 8 cm (3.1 in) of standing water over plant stems was effective. Weller (1975) also found clipping cattails too early in the growing season (e.g. May) stimulated their growth and resulted in a 25 percent increase in stem counts the following year, with an eventual decline to preclip levels. August clipping controlled up to 80 percent of cattail only if followed by submergence. It was important to remove all dead and live cattail stems to achieve this control. Cutting shoots below the water surface two or three times in one growing season before flower production reduced a cattail stand by 95-99 percent in Montana and Utah (Stodola 1967). Similar results were demonstrated by Shekhov (1974) and Sale and Wetzel (1983).

When shoots are cut below the water level, nearly all the oxygen is consumed in a short time, necessitating anaerobic respiration. In Typha, ethanol is produced accompanied by tissue breakdown after an oxygen shortage. Typha is ill adapted to deprivation of oxygen. Cuttings later than flowering stage are effective only in preventing regrowth for that year and may have no effect on subsequent years (Shekhov 1974).

Cattail control by injuring developing rhizomes and shoots was investigated (Weller 1975). Crushing and reflooding showed that cattails injured after June had poor recoveries. Success of crushing depended on the load used, number of times an area was crushed, and standing water depths after treatment. Spring and early summer treatments generally created favorable seedbeds for cattail and required a fall crushing to control seedlings. Crushing involved pulling a 55 gallon water filled drum behind a tractor. Deeper water areas showed highest control (up to 100 percent) while regrowth occurred in shallow areas. Although not practical for natural areas management, discing (Weller 1975) and blasting (Nelson and Dietz 1966) have also been investigated as methods of cattail control.

SHADING:
Black polyethylene tarps were used to cover cattails in an attempted control measure (Nelson and Dietz 1966). Actively growing cattail tips were killed when completely covered for at least sixty days. Greatest control was achieved in July when food resources of cattail were presumed to be lowest (Linde et al. 1976). Problems with holding tarps down and their degradation confounded this investigation. Cattail is generally not shade tolerant.

**Wisconsin Department of Natural Resources - Non- Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Water Level: The control of cattails by the manipulation of water level must be timed to the annual cycle of carbohydrate storage. Special leaf and stem cells called aerenchyma provide air passage from both living and dead leaves to the rhizomes. Removing dead leaves and submerging the shoots in early spring will strain the plant and eventually kill it. The depth of water necessary to kill the plants depends on temperature, the quantity of starch the plant stored the previous year, and the general vigor of the plants. Therefore, no minimum water depth can be prescribed, but, generally, a water level maintained at 3-4 feet above the tops of existing spring shoots will retard growth. It is critical to remember that even if dead leaves from the previous year are completely removed, aerobic conditions will be restored to the rhizome as soon as the new growing shoot penetrates the water surface. Even if water levels are sustained at only a few inches above the tops of the growing shoots, oxygen is prevented from reaching the rhizomes. The use of water is most efficient if the water level is raised progressively, so that all plant parts remain submerged by no more than a few inches. Water levels in the range of four to five feet also favor the wintertime survival of muskrats in flooded areas. Population levels of ten muskrats per acre, when combined with high springtime water levels, can nearly eliminate the emergence of cattails within a span of two years.

Cutting, Crushing, Shearing, and Discing: Starch reserves in the rhizomes are at their minimum in late spring when the pistillate spike of the cattail is lime green and the staminate spike is dark green. This is the best time to employ cutting, crushing, shearing, and/or discing to eliminate cattail colonies because all these methods impede starch storage during the growing season. The methods of control work best if employed during a three-week time window beginning one week before and ending one week after the staminate spike has emerged. Deep discing can retard shoot formation and damage the rhizomes, but should be used in combination with water-level control and the prevention of seed establishment to effectively hinder the re-emergence of cattails. Discing combined with continued drying and freezing in fall decreases plant survival; if a wetland can be kept dry enough to repetitively disk for 2-3 successive seasons, cattails can be eliminated or their stem densities severely reduced. However, discing has some major drawbacks: the equipment and personnel needed to carry out this method of control are costly, and will seriously disturb the site. This will likely result in the loss of other native plants in the area as well.

Cutting, crushing, shearing, or discing severs the aerenchyma link that provides oxygen between the rhizomes and leaves of cattails during dormancy. These techniques must be combined with high springtime water levels in order to effectively retard plant growth. Cattails can be cut with a rotary mower or sheared with a front-end loader on a tractor when equipment can be driven on ice, but airborne seeds may clog equipment. High water levels must be maintained throughout the spring and early summer.

Bulldozer: Bulldozers can effectively remove plants from a marsh, but will generally drastically disturb the wetland. Permits must be obtained before clearing a marsh with heavy equipment. A bulldozer or or other machinery is the only viable method that will remove floating cattail mats, but these removal methods are also costly, and effects may be short-lived. If the seed bank of the marsh is dominated by cattails, a new colony of the hardy plants may spring up after the next drawdown of the marsh; other undesirable plants could also take the place of the cattails in the marsh.

## GRAZING:

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Grazing by cows, geese, muskrats, and other animals can be an effective method of cattail management. Grazing on seedlings and young cattails without extensive rhizomes can reduce the stem density of the colony. For mature plants, grazing combined with water-level management reduces survival rates. To maximize the impact of grazing, it should be heaviest during the three-week window of time when the flower spikes are emerging.

## PRESCRIBED FIRE:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Typha spp. - http://tncweeds.ucdavis.edu/esadocs/typh_spp.html - Author: K. Motivans and S. Apfelbaum - October 27, 1987.**

Fire alone was found to provide little or no cattail control (Nelson and Dietz 1966). Fires that destroyed cattail roots offered control; however, most fires only burned above-ground biomass and did little to control cattail. Drying in readiness for burning was effective cattail control when done for two years in arid Utah. Water was pumped from wetlands and then cattail stands were allowed to sun dry.

Water level drawdown, burning (Spring, Fall, and mid-growing season), and reflooding to 20-35 cm (8-18 in) water depth or deeper controlled cattail. Fire was found useful for cattail litter cleanup and assisted access for mowing or hand clipping (Nelson and Dietz 1966, Weller 1975, Mallik and Wein 1985).

**Wisconsin Department of Natural Resources, Non-Native Plants Fact Sheets, Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Most cattail marshes must be burned in winter or before significant growth has occurred in spring; these are generally the only times when fuels are dry enough to carry a fire, although frozen ground or saturated soil may impede the fire's progress through the cattail duff. Fire is most effective as a control method when followed by naturally or artificially high water levels in the spring to smother residual stalks.

During times of drought, cattail stands overlying well-developed peat soils can be eliminated by burning. Because such fires burn peat, the ability to smother the fire by reflooding the marsh must exist before a prescribed burn can be implemented. Peat fires can also cause undesirable changes in the marsh environment, such as destruction of the seed bank, loss of peat, and air pollution.

## BIOCONTROL:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Currently there are no biological control methods for cattails.

## CHEMICAL CONTROL*:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Wick and foliar applications of systemic herbicides such as Accord®, Rodeo® or Glypro® followed by manual clipping and removal of stems can be successful. Re-treatments are usually necessary due to the extensive root system.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

For designated preserves or natural areas, especially where system-orientated stewardship is used, chemical applications may not be appropriate. This is particularly true because cattail is an element of certain natural communities. However, use of chemicals to control an overabundance of cattail may have certain applications. Spraying Dalpan (Nelson and Dietz 1966) at 8.8-35.3 kg/acre (4-16 lb/acre) produced 74-97 percent reductions in cattails ten months after a mowed area was sprayed. Cattail regrowth was sprayed at 58-90 cm (24-36 in) height in September. Control was most effective when treated areas could be flooded to 10-15 cm (4-5 in) or deeper. Dalpan spray achieved varied success but greatest control occurred where cattail stems were cut below water depths regardless of the herbicide quantity used. Poorest results were attained in areas with shallow fluctuating water levels. Spraying mature cattails rather than regrowth after cutting gave better results. Weller (1975) had similar results with spraying where Amitrol, Rodopan, and Douupon herbicides were effective in creating and maintaining openings for at least three years after spraying, but areas were quickly invaded by peripheral cattail. High doses of MCPA or 2,4-D in diesel oil (2.2-4.5 kg per acre) were effective if applied during flowering. Dalpan (9 kg/acre) and Amino-triazole (.91-1.36 kg/acre) gave good control results in Montana (Timmons et al. 1963). Herbicide applications were found necessary for up to three years in some areas. Similar results were found by Grigsby et

al. (1955), Heath and Lewis (1957), Krolikowska (1976), Pahuja et al. (1980), Singh and Moolani (1973), and Wisconsin Department of Natural Resources (1969).

Wick and spray applications of Roundup followed by manual clipping of all cattail stems was the treatment conducted by Applied Ecological Services and All Services Company (1985) at a pond in northern Illinois. Cattail seeds were just at ripening stage at the time of treatment. Retreatment of Roundup several weeks later and subsequent die-off proved this method successful.

Herbicide treatment at flowering may stress the cattail plants more than at other stages since the energy investment by the plant has been channeled into flowering.

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Application in mid to late summer enhances the effectiveness of translocated herbicides, although the herbicides will have little effect on seed production during the year of application. A hemi-marsh may be created if some cattails survive, although the ability of the marsh to persist in this condition depends on the manipulation of water levels. Water level control to minimize recruitment from the seed bank must be used to ensure cattails will not return once reduced by herbicides.

Herbicides can be detrimental to wetlands habitats--be sure to use herbicides that readily break down in water, soil, or substrate, such as glyphosate formulated for use over water. Boom or wick applications by ground or air boat are best for small areas where pesticide drift is a concern. Aerial applications may be used on large areas. Herbicidal control of cattails may be costly, although actual application of the herbicide usually represents a small fraction of this cost. One area manager found that an aerial application of glyphosate during August was effective in controlling cattails, dogwood, and willow, but quite costly at $110/acre. Due to the possibility of fish contamination, notice must be posted before spraying, and can be done only by a person licensed to apply herbicides.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

# Narrow-leaved and Hybrid Cattail
## *Typha angustifolia* and *T. x glauca*

**SOURCE**: The following information on this species is taken, in part, from the Ohio Department of Natural Resources, Division of Natural Areas and Preserves, Invasive Plants of Ohio Fact Sheets website at http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is January 8, 2003.

**DESCRIPTION:** Narrow-leaved cattail is a non-native, invasive plant that hybridizes with the native broad-leaved cattail (*T. latifolia*) to produce the invasive *T. xglauca*. All three aquatic perennials may grow up to a height of 10 feet and produce a velvety brown spike of flowers. The flower head of the hybrid and the narrow-leaved cattail have a gap of 1-4 inches between the male and female flowers, while the native species has both flower types next to each other. The leaves of cattail originate from the base and spread outward. The narrow-leaved and hybrid cattails have leaves that are 1/4 - 3/4 inch across; the native cattail's leaves are wider at 1/2 -1 inch. A starchy rhizome forms beneath each plant.



Narrow-leaved (Left) and Broad-leaved (Right) Cattail Division Photo

Photo: Ohio Department of Natural Resources, Division of Natural Areas and Preserves, http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm



Photo of T. angustifolia: © Mandy Tu/ The Nature Conservancy

**HABITAT**: Stands of cattail can be found in a wide variety of wetland habitats, including marshes, lakeshores, river backwaters and roadside ditches. This prolific plant can grow in disturbed areas, as well as brackish, and polluted waters of depths nearing 3 feet.

**DISTRIBUTION:** Narrow-leaved cattails are believed to have been introduced to the Atlantic seaboard from the dry ballast of European ships. This plant has since spread westward and occurs throughout much of the United States. The hybrid cattail is concentrated in the northeast, but may occur wherever both the native and the narrow-leaved species are present. All three taxa are found throughout Ohio.

**PROBLEM:** Narrow-leaved and hybrid cattail will out-compete native plants in wetland systems. These plants establish dense monocultures that enable them to shade out native vegetation. They are also thought to be allelopathic, producing chemicals which discourage growth of other plant species. Cattails reproduce both vegetatively by rhizomes and sexually through massive amounts of seed.

Photo: © Mandy Tu/ The Nature Conservancy



**For more information on non-native cattails, please contact:**

Grace, J.B. and J.S. Harrison. 1986. The Biology of Canadian Weeds: Typha latifolia L., T. angustifolia L. and T. xglauca Godr. Canadian Journal of Plant Science 66: 361-379.
Hoffman, R. and K. Kearns, eds. 1997. Wisconsin Manual of Control Recommendations for Ecologically Invasive Plants. Bureau of Endangered Resources, Wisconsin Department of Natural Resources.
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
Motivans, K. and S. Apfelbaum. 1987. Element Stewardship Abstract for Typha spp.
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Ohio Department of Natural Resources, Division of Natural Areas and Preserves, http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
The Nature Conservancy, Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm
Wisconsin Department of Natural Resources, Non-native plants fact sheets, Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm

**Management and Control Information**

Return to List




## Species Management and Control Information

# Narrow-leaved and Hybrid Cattail
# *Typha angustifolia* and *T. x glauca*

**SOURCE**: The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

The acreage of cattail-dominated wetlands in the United States has increased drastically since the early twentieth century due to changes in hydrology and land use. The optimal control technique for a given site will depend on the hydrologic state of the site, the size of the area to be managed, and if the manager is able to manipulate water levels.

## MANUAL AND MECHANICAL CONTROL:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Manipulation of water levels can kill cattails by inhibiting airflow from the cattail shoots to the roots. Removing the dead leaves and submerging the shoots in early spring will eliminate gas diffusion and "suffocate" the plant. In situations where water level manipulations are either not feasible or appropriate, pulling, cutting and bulldozing treatments have been used with some success. In the case of bulldozing, the benefits in effective removal may not outweigh the costs of disturbing the wetland.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Typha spp. - http://tncweeds.ucdavis.edu/esadocs/typh_spp.html - Author: K. Motivans and S. Apfelbaum - October 27, 1987.**

WATER LEVEL MODIFICATION:
High water conditions in a cattail stand can affect the growth of seedlings, can break off mature stalks, or can be followed by the immigration of muskrats which eat the cattail (Zimmerman pers. comm.). The effect of flooding does not always have negative impacts on cattails -- plants have been known to float up and continue growing until water returns to a previous lower levels.

As with any control measure, temporary conditions, such as flooding, do not prevent later seed establishment. Cattail seeds can arrive from a great distance, and it doesn't take but a few seeds to germinate and rapidly produce clones as adults. The cost of management actions should be considered when dealing with unknown response variables.

Low water conditions, maintained by draining a wetland, significantly effects the overall community (Mallik and Wein 1985). Harris and Marshall (1963) concluded that draining techniques have possible detrimental effects because the plant composition of a wetland can be radically changed. Draining alone can cause a significant increase in Typha cover under some conditions (Mallik and Wein 1985). However, to inhibit Typha growth, a wetland can be drained and then burned during the summer. If there is no reserve of water over winter cattails will not survive the following spring, according to Zimmerman (pers. comm.) but there have been no controlled experiments to show this.

Two years of 65 cm (26 in) deep flooding was required before established cattail began to die and open water conditions were created at

Sinnissippi Marsh. Cattail initially survived flooding from 1973-1977 and became the dominant emergent plant. A light green color, noticeably narrower leaves, and absence of fruiting heads indicated stress in 1976. Cattail stem densities declined 57 percent with all emergent plants dead in 1977. Horicon Marsh, flooded to a depth of 40 cm (16 in), showed declines in emergent and aquatic plants. Cattail required two years before it declined (Wisconsin DNR 1969 and 1971).

Mature T. latifolia and seedlings less than one year old are killed by water depths of 63.5 cm (25 in) and 45 cm (18 in) or more, respectively. Narrow-leaved cattail was unaffected by this degree of flooding. Narrow-leaved cattail establishment was prevented when water levels were maintained at 1.2 m (47 in) or deeper (Steenis et al. 1958). Dryer conditions allowed more clones of T. angustifolia to be spread (McMillan 1959).

Because cattails can transpire significant quantities of water (2-3m of water/acre/year) (Fletcher and Elmendor 1955, Zohary 1962), their establishment may serve to exacerbate water level instability and further contribute to disruptive influences supporting increased cattail. Flooding must account for evapotranspirational losses of water to maintain a level effective in cattail control.

PHYSICAL CONTROL:
Hand or mechanical cutting of cattails followed by submergence of all cattail stems results in high control. Up to 100 percent cattail control was measured two growing seasons after treatment. No visible cattail regrowth occurred in one year and cattail rhizomes were dead. The highest cattail control of any method tested was achieved by two clippings followed by stem submergence to at least 7.5 cm (3 in) (Nelson and Dietz 1966). Control was best if plants were cut in late summer or early fall.

In Iowa (Weller 1975), cutting cattail and reflooding with at least 8 cm (3.1 in) of standing water over plant stems was effective. Weller (1975) also found clipping cattails too early in the growing season (e.g. May) stimulated their growth and resulted in a 25 percent increase in stem counts the following year, with an eventual decline to preclip levels. August clipping controlled up to 80 percent of cattail only if followed by submergence. It was important to remove all dead and live cattail stems to achieve this control. Cutting shoots below the water surface two or three times in one growing season before flower production reduced a cattail stand by 95-99 percent in Montana and Utah (Stodola 1967). Similar results were demonstrated by Shekhov (1974) and Sale and Wetzel (1983).

When shoots are cut below the water level, nearly all the oxygen is consumed in a short time, necessitating anaerobic respiration. In Typha, ethanol is produced accompanied by tissue breakdown after an oxygen shortage. Typha is ill adapted to deprivation of oxygen. Cuttings later than flowering stage are effective only in preventing regrowth for that year and may have no effect on subsequent years (Shekhov 1974).

Cattail control by injuring developing rhizomes and shoots was investigated (Weller 1975). Crushing and reflooding showed that cattails injured after June had poor recoveries. Success of crushing depended on the load used, number of times an area was crushed, and standing water depths after treatment. Spring and early summer treatments generally created favorable seedbeds for cattail and required a fall crushing to control seedlings. Crushing involved pulling a 55 gallon water filled drum behind a tractor. Deeper water areas showed highest control (up to 100 percent) while regrowth occurred in shallow areas. Although not practical for natural areas management, discing (Weller 1975) and blasting (Nelson and Dietz 1966) have also been investigated as methods of cattail control.

SHADING:
Black polyethylene tarps were used to cover cattails in an attempted control measure (Nelson and Dietz 1966). Actively growing cattail tips were killed when completely covered for at least sixty days. Greatest control was achieved in July when food resources of cattail were presumed to be lowest (Linde et al. 1976). Problems with holding tarps down and their degradation confounded this investigation. Cattail is generally not shade tolerant.

**Wisconsin Department of Natural Resources - Non- Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Water Level: The control of cattails by the manipulation of water level must be timed to the annual cycle of carbohydrate storage. Special leaf and stem cells called aerenchyma provide air passage from both living and dead leaves to the rhizomes. Removing dead leaves and submerging the shoots in early spring will strain the plant and eventually kill it. The depth of water necessary to kill the plants depends on temperature, the quantity of starch the plant stored the previous year, and the general vigor of the plants. Therefore, no minimum water depth can be prescribed, but, generally, a water level maintained at 3-4 feet above the tops of existing spring shoots will retard growth. It is critical to remember that even if dead leaves from the previous year are completely removed, aerobic conditions will be restored to the rhizome as soon as the new growing shoot penetrates the water surface. Even if water levels are sustained at only a few inches above the tops of the growing shoots, oxygen is prevented from reaching the rhizomes. The use of water is most efficient if the water level is raised progressively, so that all plant parts remain submerged by no more than a few inches. Water levels in the range of four to five feet also favor the wintertime survival of muskrats in flooded areas. Population levels of ten muskrats per acre, when combined with high springtime water levels, can nearly eliminate the emergence of cattails within a span of two years.

Cutting, Crushing, Shearing, and Discing: Starch reserves in the rhizomes are at their minimum in late spring when the pistillate spike of the cattail is lime green and the staminate spike is dark green. This is the best time to employ cutting, crushing, shearing, and/or discing to eliminate cattail colonies because all these methods impede starch storage during the growing season. The methods of control work best if employed during a three-week time window beginning one week before and ending one week after the staminate spike has emerged. Deep discing can retard shoot formation and damage the rhizomes, but should be used in combination with water-level control and the prevention of seed establishment to effectively hinder the re-emergence of cattails. Discing combined with continued drying and freezing in fall decreases plant survival; if a wetland can be kept dry enough to repetitively disk for 2-3 successive seasons, cattails can be eliminated or their stem densities severely reduced. However, discing has some major drawbacks: the equipment and personnel needed to carry out this method of control are costly, and will seriously disturb the site. This will likely result in the loss of other native plants in the area as well.

Cutting, crushing, shearing, or discing severs the aerenchyma link that provides oxygen between the rhizomes and leaves of cattails during dormancy. These techniques must be combined with high springtime water levels in order to effectively retard plant growth. Cattails can be cut with a rotary mower or sheared with a front-end loader on a tractor when equipment can be driven on ice, but airborne seeds may clog equipment. High water levels must be maintained throughout the spring and early summer.

Bulldozer: Bulldozers can effectively remove plants from a marsh, but will generally drastically disturb the wetland. Permits must be obtained before clearing a marsh with heavy equipment. A bulldozer or or other machinery is the only viable method that will remove floating cattail mats, but these removal methods are also costly, and effects may be short-lived. If the seed bank of the marsh is dominated by cattails, a new colony of the hardy plants may spring up after the next drawdown of the marsh; other undesirable plants could also take the place of the cattails in the marsh.

## GRAZING:

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Grazing by cows, geese, muskrats, and other animals can be an effective method of cattail management. Grazing on seedlings and young cattails without extensive rhizomes can reduce the stem density of the colony. For mature plants, grazing combined with water-level management reduces survival rates. To maximize the impact of grazing, it should be heaviest during the three-week window of time when the flower spikes are emerging.

## PRESCRIBED FIRE:

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Typha spp. - http://tncweeds.ucdavis.edu/esadocs/typh_spp.html - Author: K. Motivans and S. Apfelbaum - October 27, 1987.**

Fire alone was found to provide little or no cattail control (Nelson and Dietz 1966). Fires that destroyed cattail roots offered control; however, most fires only burned above-ground biomass and did little to control cattail. Drying in readiness for burning was effective cattail control when done for two years in arid Utah. Water was pumped from wetlands and then cattail stands were allowed to sun dry.

Water level drawdown, burning (Spring, Fall, and mid-growing season), and reflooding to 20-35 cm (8-18 in) water depth or deeper controlled cattail. Fire was found useful for cattail litter cleanup and assisted access for mowing or hand clipping (Nelson and Dietz 1966, Weller 1975, Mallik and Wein 1985).

**Wisconsin Department of Natural Resources, Non-Native Plants Fact Sheets, Common cattail and Narrow-leaved cattail, http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Most cattail marshes must be burned in winter or before significant growth has occurred in spring; these are generally the only times when fuels are dry enough to carry a fire, although frozen ground or saturated soil may impede the fire's progress through the cattail duff. Fire is most effective as a control method when followed by naturally or artificially high water levels in the spring to smother residual stalks.

During times of drought, cattail stands overlying well-developed peat soils can be eliminated by burning. Because such fires burn peat, the ability to smother the fire by reflooding the marsh must exist before a prescribed burn can be implemented. Peat fires can also cause undesirable changes in the marsh environment, such as destruction of the seed bank, loss of peat, and air pollution.

## BIOCONTROL:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Currently there are no biological control methods for cattails.

## CHEMICAL CONTROL*:

**Ohio Department of Natural Resources - Division of Natural Areas and Preserves - Invasive Plants of Ohio Fact Sheet #11 - Narrow-leaved and Hybrid Cattail - http://www.dnr.state.oh.us/dnap/invasive/11cattail.htm - January 8, 2003.**

Wick and foliar applications of systemic herbicides such as Accord®, Rodeo® or Glypro® followed by manual clipping and removal of stems can be successful. Re-treatments are usually necessary due to the extensive root system.

**The Nature Conservancy - Invasive Species Initiative - Species Management Summary (ESA or Element Stewardship Abstract): Alliaria petiolata - http://tncweeds.ucdavis.edu/esadocs/allipeti.html - Author: Victoria Nuzzo - February 2000.**

For designated preserves or natural areas, especially where system-orientated stewardship is used, chemical applications may not be appropriate. This is particularly true because cattail is an element of certain natural communities. However, use of chemicals to control an overabundance of cattail may have certain applications. Spraying Dalpan (Nelson and Dietz 1966) at 8.8-35.3 kg/acre (4-16 lb/acre) produced 74-97 percent reductions in cattails ten months after a mowed area was sprayed. Cattail regrowth was sprayed at 58-90 cm (24-36 in) height in September. Control was most effective when treated areas could be flooded to 10-15 cm (4-5 in) or deeper. Dalpan spray achieved varied success but greatest control occurred where cattail stems were cut below water depths regardless of the herbicide quantity used. Poorest results were attained in areas with shallow fluctuating water levels. Spraying mature cattails rather than regrowth after cutting gave better results. Weller (1975) had similar results with spraying where Amitrol, Rodopan, and Douupon herbicides were effective in creating and maintaining openings for at least three years after spraying, but areas were quickly invaded by peripheral cattail. High doses of MCPA or 2,4-D in diesel oil (2.2-4.5 kg per acre) were effective if applied during flowering. Dalpan (9 kg/acre) and Amino-triazole (.91-1.36 kg/acre) gave good control results in Montana (Timmons et al. 1963). Herbicide applications were found necessary for up to three years in some areas. Similar results were found by Grigsby et

al. (1955), Heath and Lewis (1957), Krolikowska (1976), Pahuja et al. (1980), Singh and Moolani (1973), and Wisconsin Department of Natural Resources (1969).

Wick and spray applications of Roundup followed by manual clipping of all cattail stems was the treatment conducted by Applied Ecological Services and All Services Company (1985) at a pond in northern Illinois. Cattail seeds were just at ripening stage at the time of treatment. Retreatment of Roundup several weeks later and subsequent die-off proved this method successful.

Herbicide treatment at flowering may stress the cattail plants more than at other stages since the energy investment by the plant has been channeled into flowering.

**Wisconsin Department of Natural Resources - Non-Native Plants Fact Sheets - Common cattail and Narrow-leaved cattail - http://www.dnr.state.wi.us/org/land/er/invasive/factsheets/cattail.htm**

Application in mid to late summer enhances the effectiveness of translocated herbicides, although the herbicides will have little effect on seed production during the year of application. A hemi-marsh may be created if some cattails survive, although the ability of the marsh to persist in this condition depends on the manipulation of water levels. Water level control to minimize recruitment from the seed bank must be used to ensure cattails will not return once reduced by herbicides.

Herbicides can be detrimental to wetlands habitats--be sure to use herbicides that readily break down in water, soil, or substrate, such as glyphosate formulated for use over water. Boom or wick applications by ground or air boat are best for small areas where pesticide drift is a concern. Aerial applications may be used on large areas. Herbicidal control of cattails may be costly, although actual application of the herbicide usually represents a small fraction of this cost. One area manager found that an aerial application of glyphosate during August was effective in controlling cattails, dogwood, and willow, but quite costly at $110/acre. Due to the possibility of fish contamination, notice must be posted before spraying, and can be done only by a person licensed to apply herbicides.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Fiveleaf Akebia
## Akebia quinata (Houtt.) Dcne.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is April, 2004.

**NATIVE RANGE**: Central China to Korea and Japan

**DESCRIPTION**: Akebia quinata, also known as chocolate vine, is a twining woody vine in the mostly tropical Lardizabalaceae family. It grows as either a twining vine or vigorous groundcover and has slender, rounded stems that are green when young and brown at maturity. The palmate (like a hand) leaves of akebia alternate along the stem and are divided into five, or sometimes fewer, approximately equal parts called leaflets, whose small stems meet at a central juncture. Leaflets are generally long oval in shape, 1½ to 3 inches long and emerge with a purplish tinge, becoming blue-green at maturity.




Photo: Shep Zedaker, Virginia Polytechnic Institute and State University, www.forestryimages.org.

Flowers are unusual, chocolate-purple colored, and fragrant, about 1 inch across, and appear in late March to early April, when they are often concealed by new foliage. Fruits are purple-violet, flattened sausage-like pods, 2 1/4 to 4 inches in length, that ripen in late September to early October. The inside of the pod has a whitish pulpy core with many tiny black seeds. Fiveleaf akebia is deciduous in cooler climates but may remain evergreen in the warmer regions, such as Louisiana.

**ECOLOGICAL THREAT**: Fiveleaf akebia grows so quickly that, if left unmanaged, it can kill off existing ground level vegetation, understory shrubs and trees, and even some canopy trees, by overtopping and smothering them. Once established, its dense growth prevents seed germination and seedling establishment of native plants.




Photo: Shep Zedaker, Virginia Polytechnic Institute and State University, www.forestryimages.org.

**DISTRIBUTION IN THE UNITED STATES**: Fiveleaf akebia is found in 16 states in the eastern U.S., from Michigan to Connecticut, south to Georgia. Although it is not listed as occurring in South Carolina, this is probably an oversight.

**HABITAT IN THE UNITED STATES**: Akebia is shade and drought tolerant and can invade many types of habitats. Its growth appears to be restricted only by the height of the object it is entangling.

**BACKGROUND**: Fiveleaf akebia brought to the United States in 1845 as an ornamental and has since naturalized in the warmer climates.

**BIOLOGY & SPREAD**: Akebia spreads primarily by vegetative means and is capable of growing twenty to forty feet in a single growing season. Fruits are not always produced and the seeds of akebia are not known to be carried by wind or insects. While birds may play a role in seed dispersal, fiveleaf akebia is vectored primarily through inadvertent activities of humans.

**SUGGESTED ALTERNATIVE PLANTS**: Some native alternative vines for the eastern U.S. include trumpet honeysuckle (Lonicera sempervirens), cross vine (Bignonia capreolata), trumpet creeper (Campsis radicans), or Dutchman's pipe (Aristolochia durior). Contact your local native plant society for other suggestions.

**AUTHORS**:
Adrienne Reese and Robert E. Lyons, Dept. of Horticulture, Virginia Tech, Blacksburg, VA. Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**EDITOR**:
Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on garlic mustard, please contact:**
The Bugwood Network, http://www.invasive.org/.

**REFERENCES:**
Dirr, Michael. Manual of Woody Landscape Plants. Illinois: Champaign, 1990. DeWolf, Gordon, et al. Taylor's Guide to Ground Covers. Boston: Houghton Mifflin, 1987. Wyman, Donald. Shrubs and Vines for American Gardens. Toronto: The Macmillan Company, 1969.

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Back to List




## Species Management and Control Information

# Fiveleaf Akebia
# *Akebia quinata* (Houtt.) Dcne.

**SOURCE:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resources:

**Plant Conservation Alliance, Alien Plant Working Group - Weed Gone Wild - Fact sheet - Fiveleaf Akebia - http://www.nps.gov/plants/alien/fact/akqu1.htm - Authors: Adrienne Reese, Robert E. Lyons and Jil M. Swearingen - April 1, 2004.**

## MANUAL AND MECHANICAL CONTROL:

Small infestations of fiveleaf akebia can be controlled mechanically. Because of its extremely rapid growth, cutting will need to be repeated throughout the season. At a minimum, akebia should be cut back to the ground at the end of the summer. Akebia vines may also be dug up, removing as much of the roots as possible. To ensure its complete removal, regular monitoring and repeated cutting, digging or pulling is necessary.

## CHEMICAL CONTROL:

For large infestations, use of a labelled systemic herbicide, such as glyphosate (e.g., Roundup) or triclopyr (e.g., Garlon), is probably the most effective method to control akebia. An herbicidal soap, such as pelargonic acid (e.g., Scythe), which provides a burndown of plant tissues, may also provide some control. The latter product may also provide enhanced uptake of certain systemic herbicides, such as glyphosate. Be sure to avoid drift onto or direct spray of desirable plant species during herbicide applications

**DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers**

## Porcelain-berry
## Ampelopsis brevipedunculata (Maxim.) Trautv.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is February,, 2005.

**NATIVE RANGE**: Northeast Asia - China, Korea, Japan, and Russian Far East

**DESCRIPTION**: Porcelain-berry is a deciduous, woody, perennial vine in the grape family (Vitaceae).



Photo: Jil Swearingen, USDI National Park Service

It twines with the help of non-adhesive tendrils that occur opposite the leaves and closely resembles native grapes in the genus Vitis. The stem pith of porcelain-berry is white (grape is brown) and continuous across the nodes (grape is not), the bark has lenticels (grape does not), and the bark does not peel (grape bark peels or shreds). The leaves are alternate, broadly ovate with a heart-shaped base, palmately 3-5 lobed or more deeply dissected, and have coarsely toothed margins. The inconspicuous, greenish-white flowers with "free" petals occur in cymes opposite the leaves from June through August (in contrast to grape species that have flowers with petals that touch at tips and occur in panicles. The fruits appear in September-October and are colorful, changing from pale lilac, to green, to a bright blue. Porcelain-berry is often confused with species of grape (Vitis) and may be confused with several native species of Ampelopsis -- Ampelopsis arborea and Ampelopsis cordata.

**ECOLOGICAL THREAT**: Porcelain-berry is a vigorous invader of open and wooded habitats. It grows and spreads quickly in areas with high to moderate light. As it spreads, it climbs over shrubs and other vegetation, shading out native plants and consuming habitat.



Photo: Jil Swearingen,
USDI National Park Service, www.invasive.org.

**DISTRIBUTION IN THE UNITED STATES**: Porcelain-berry is found from New England to North Carolina and west to Michigan (click here for USDA Plants map) and is reported to be invasive in twelve states in the Northeast: Connecticut, Delaware, Massachusetts, Maryland, New Jersey, New York, Pennsylvania, Rhode Island, Virginia, Washington D.C., West Virginia, and Wisconsin. Click here to see a distribution map.

**HABITAT IN THE UNITED STATES**: Porcelain-berry grows well in most soils, especially forest edges, pond margins, stream banks, thickets, and waste places, where there is full sunlight to partial shade, and where it is not permanently wet. Porcelain-berry appears to be less tolerant of heavily shaded areas, such as that found in mature forest interiors.

**BACKGROUND**: Porcelain-berry was originally cultivated around the 1870s as a bedding and landscape plant. In spite of its aggressiveness in some areas, it is still used in the horticultural trade (for example, the ornamental A. brevipedunculata 'Elegans' is often recommended as a landscape plant with a cautionary note that "care must be taken to keep it from overtaking and shading out small plants"). The same characteristics that make porcelain-berry a desirable plant for the garden -- its colorful berries, good ground coverage, trellis-climbing vines, pest-resistance, and tolerance of adverse conditions -- are responsible for its presence in the United States as an undesirable invader.

**BIOLOGY & SPREAD**: Porcelain-berry spreads by seed and through vegetative means. The colorful fruits, each with two to four seeds, attract birds and other small animals that eat the berries and disperse the seeds in their droppings. The seeds of porcelain-berry germinate readily to start new infestations. Porcelain-berry is often found growing in riparian areas downstream from established patches, suggesting they may be dispersed by water also. The taproot of porcelain-berry is large and vigorous. Resprouting will occur in response to cutting of above-ground portions.

**For more information on Porcelainberry, please contact:**
Lisa Jameson, National Park Service, National Capital Region, Exotic Plant Management Team, Washington, DC, 20007;
Lisa_Jameson@nps.gov.
Susan Salmons, US National Park Service, Rock Creek Park, Washington, DC; Sue_Salmons at nps.gov; 202-426-6834, ext. 33
Jil Swearingen, US National Park Service, Center for Urban Ecology, Washington, DC; Jil_Swearingen@nps.gov.
Virginia Natural Heritage Program Fact Sheet--Garlic Mustard; http://www.dcr.virginia.gov/dnh/invlist.htm
The Nature Conservancy Invasive Species Initiative, http://tncweeds.ucdavis.edu/esadocs.html
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov

**SUGGESTED ALTERNATIVE PLANTS**: Many lovely non-invasive vines are available. Some native substitutes to consider include trumpet honeysuckle (Lonicera sempervirens), trumpet creeper (Campsis radicans), American wisteria (Wisteria frutescens)*, Virginia creeper (Parthenocissus quinquefolia), and goldflame honeysuckle (Lonicera heckrottii). In the southeast, several species of native Ampelopsis occur and should be considered if the habitat is appropriate. Please consult the native plant society in your state for more suggestions and information on sources of native plants.

* If you wish to plant wisteria, make certain that it is the native species. Two commonly planted ornamental wisterias, Chinese wisteria (Wisteria sinensis) and Japanese wisteria (Wisteria floribunda), are exotic and aggressive invaders.

**AUTHOR**: Jamie Young, National Research Council, Washington, DC.
**EDITOR**: Jil Swearingen, U.S. National Park Service, Washington, DC.
**REVIEWERS**: Carol Jelich, Ann F. Rhoads, and Louisa Thompson.
**REFERENCES:**
Dirr, Michael A. 1998. Manual of Woody Landscape Plants. Stipes Publishing, Chicago.
Gleason, H.A. and Cronquist, A. 1991. Manual of Vascular Plants of Northeastern United States and Adjacent Canada, Second Edition. The New York Botanical Garden, Bronx, NY .
Magee, D.W. and H.E. Ahles. 1999. Flora of the Northeast. University of Massachusetts Press, Amherst.
Maryland Native Plant Society. Control of Invasive Non-Native Plants: A Guide for Gardeners and Homeowners in the Mid-Atlantic Region. Online. Available: http://mdflora.org/publications/invasives.htm
Randall, J. M., and Marinelli, J. 1996. Invasive Plants: Weeds of the Global Garden. Brooklyn Botanic Garden, Handbook #149.
Rehder, A. Manual of Cultivated Trees and Shrubs Hardy in North America Exclusive of the Subtropical and Warmer Temperate Zones, 2nd ed. The MacMillan Company, New York. 996 pp.
Rhoads, A.F. and T.A Block. 2000.The Plants of Pennsylvania, An Illustrated Manual. University of Pennsylvania Press. 1061 pp.
Robertson, D.J., M.C. Robertson, and T. Tague. 1994. Colonization dynamics of four exotic plants in a northern Piedmont natural area. Bulletin of the Torrey Botanical Club 121(2):107-118.
Rose, N. 1998. Field Notes: Ampelopsis brevipedunculata 'Elegans'. American Nurseryman.
Salmons, S. 2000. Rock Creek Park Invasive Non-Native Plant Mitigation Program. Final Report. National Park Service, Rock Creek Park, Washington, DC.
Swearingen, J. 2004. WeedUS: Database of Invasive Plants of Natural Areas in the U.S. Plant Conservation Alliance.
http://www.nps.gov/plants/alien USDA, ARS, National Genetic Resources Program. Germplasm Resources Information Network - (GRIN) [Online Database]. National Germplasm Resources Laboratory, Beltsville, Maryland. URL: http://www.ars-grin.gov/var/apache/cgi-bin/npgs/html/taxon.pl?2964 (01 September 2004).
Virginia Native Plant Society. Invasive Alien Plant Species of Virginia: Porcelain-berry (Ampelopsis brevipedunculata (Maxim.) Trautv.).

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Porcelain-berry
# *Ampelopsis brevipedunculata* (Maxim.) Trautv.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

All information that follows in taken from the following resources:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Porcelain-berry- http://www.nps.gov/plants/alien/fact/ambr1.htm - Author: Jamie Young - February 3, 2005**

## GENERAL MANAGEMENT CONSIDERATIONS:

Because porcelain-berry vines can grow up to 15 ft. in a single growing season, especially when rainfall is abundant, and seed may be viable in the soil for several years, effective control requires dedicated followup. Treatment measures often must be repeated during the growing season and for several years afterwards to fully eradicate the plant. Prevention of flowering, fruiting and production of mature seeds will help reduce its spread.

## MANUAL AND MECHANICAL CONTROL:

Hand pulling of vines in the fall or spring will prevent flower buds from forming the following season. Where feasible, plants should be pulled up by hand before fruiting to prevent the production and dispersal of seeds. If the plants are pulled while in fruit, the fruits should be bagged and disposed of in a landfill. For vines too large to pull out, cut them near the ground and either treat cut stems with systemic herbicide or repeat cutting of regrowth as needed.

## CHEMICAL CONTROL:

Chemical control in combination with manual and mechanical methods is effective and likely to be necessary for large infestations. The systemic herbicides triclopyr (e.g., Garlon 3A and Garlon 4) and glyphosate (e.g., Roundup and Rodeo) have been used successfully by many practitioners.
Foliar applications. The most effective control has been achieved using triclopyr formulations. From summer to fall, apply a water-based solution of 2.5% Garlon 3A (triclopyr amine) to foliage or cut plants first, allow time for regrowth and then apply the mixture. Smaller infestations can be controlled to some extent with spot applications of glyphosate to leaves, used sparingly to avoid contact of desirable plants with spray. Cut the vines back during the summer and allow to resprout before applying herbicide, or apply glyphosate to leaves in early autumn, just prior to senescence.
Basal bark applications. Apply a mixture of 20-30% Garlon 4 (triclopyr ester) mixed with commercially available basal oil, horticultural oil, diesel fuel, No. 1 or No. 2 fuel oil, or kerosene, to 2 - 3 ft. long sections of stem near the base of the vines.

Return to List




## Oriental bittersweet
## Celastrus orbiculatus Thunb.

**SOURCE:** The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets. The Fact Sheet information for this species can be found at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Specific species management and control information is provided elsewhere. Check the website link provided to check for future updates. The last update for this fact sheet is March 1999.

**NATIVE RANGE**: Eastern Asia, Korea, China and Japan

**DESCRIPTION**: Oriental bittersweet is a deciduous, woody, perennial vine in the staff-tree family (Celastraceae), which sometimes occurs as a trailing shrub. Also known as round-leaved and Asiatic bittersweet, stems of older plants sometimes grow to four inches in diameter.



Photo: Jil M. Swearingen,
U.S. National Park Service, Washington, D.C.




Photo: James H. Miller,
USDA Forest Service, www.invasive.org

Leaves of oriental bittersweet are glossy, rounded, finely toothed and arranged alternately along the stem. Clusters of small greenish flowers emerge from leaf axils, allowing each plant to produce large numbers of seeds.



Photo: Leslie J. Mehrhoff,
Univ. of Connecticut, www.invasive.org.



Photo: James H. Miller,
USDA Forest Service, www.invasive.org.

At maturity, globular, green to yellow fruits split open to reveal three red-orange, fleshy arils that contain the seeds. These showy fruits have made oriental bittersweet very popular for use in floral arrangements. Since this plant is easily confused with our native climbing bittersweet vine (Celastrus scandens), which flowers at the tips rather than along the stems, it is imperative that correct identification be made before controls are attempted.

**ECOLOGICAL THREAT**: Oriental bittersweet is an aggressive invader that threatens all vegetation levels of forested and open areas. It grows

over other vegetation, completely covering it, and kills other plants by preventing photosynthesis, girdling, and uprooting by force of its massive weight. In the northeastern U.S., exotic Oriental bittersweet appears to be displacing the native climbing bittersweet, Celastrus scandens, which occurs in similar habitats, through competition and hybridization.

**DISTRIBUTION IN THE UNITED STATES**: Oriental bittersweet currently occurs from New York to North Carolina, and westward to Illinois. Click here to see a distribution map.

**HABITAT IN THE UNITED STATES**: Oriental bittersweet infests forest edges, woodlands, early successional fields, hedgerows, coastal areas and salt marsh edges, particularly those suffering some form of land disturbance. While often found in more open, sunny sites, its tolerance for shade allows oriental bittersweet to invade forested areas.

**BACKGROUND**: Introduced into the U.S. in the 1860s as an ornamental plant, oriental bittersweet is often associated with old homesites, from which it has escaped into surrounding natural areas. Oriental bittersweet is still widely planted and maintained as an ornamental vine, further promoting its spread.

**BIOLOGY & SPREAD**: Oriental bittersweet reproduces prolifically by seed, which is readily dispersed to new areas by many species of birds. Its seeds germinate in late spring in partial to dense shade. It also expands vegetatively by stolons (above-ground stems), and rhizomes (underground stems), and through root suckering, the ability to send shoots up from the roots.

**For more information on Oriental bittersweet, please contact:**
Invasive Plant Atlas of New England - University of Connecticut http://webapps.lib.uconn.edu/ipane/browsing.cfm?descriptionid=27
Kris Johnson, Great Smoky Mountains National Park, Gatlinburg TN
Nonnative Invasive Plants of Southern Forests - USDA Forest Service http://www.invasive.org/eastern/srs/OAB.html
Plant Invaders of Mid-Atlantic Natural Areas, National Park Service and U.S. Fish and Wildlife Service, http://www.invasive.org/eastern/midatlantic/ceor.html
Southeast Exotic Pest Plant Council, Invasive Plant Manual, SE-EPPC http://www.invasive.org/eastern/eppc/bittersweet.html
The Nature Conservancy, Element Stewardship Abstract - http://tncweeds.ucdavis.edu/esadocs/documnts/celaorb.html
Virginia Natural Heritage Program Fact Sheet--Oriental bittersweet http://www.state.va.us/~dcr/dnh/invcela.htm

**SUGGESTED ALTERNATIVE PLANTS**:Several attractive native vines are available that provide nectar, seed and host plant material for butterflies, hummingbirds, and other wildlife. These include common, or climbing, bittersweet (Celastrus scandens) which is native to the eastern U.S., trumpet creeper (Campsis radicans), passionflower vine (Passiflora lutea), pipevine or Dutchman's pipe (Aristolochia macrophylla) and native wisteria* (Wisteria frutescens).

*If you wish to plant wisteria, make certain that it is the native species. Two commonly planted ornamental wisterias, Chinese wisteria (Wisteria sinensis) and Japanese wisteria (Wisteria floribunda), are exotic and aggressive invaders.

**AUTHORS:**
Carole Bergmann, Montgomery County Department of Parks, Silver Spring, MD. Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**REFERENCES:**
Dirr, Michael A. 1990. Manual of wody landscape plants: their identification, ornamental characteristics, culture, propagation and uses. Stipes Publishing Company, Champaign, IL.
Harty, Francis M. 1993. How Illinois kicked the exotic species habit. Pp. 195-209. In B.N. McKnight (ed.), Biological Pollution, Indiana Acad. Sci., Indianapolis, IN. 261 pp.
Hutchinson, M. 1992. Vegetation management guideline: round-leaved bittersweet (Celastrus orbiculatus). Natural Areas Journal 12:161.
McNab, W.H. and M. Meeker. 1987. Oriental bittersweet: a growing threat to hard-wood silviculture in the Appalachians. Northern Journal of Applied Forestry 4:174-177.
Shepard, C. 1996. Invasive Plant Information Sheet: Asiatic Bittersweet (Celastrus orbiculatus Thunb.). The Nature Conservancy, Connecticut Chapter, Hartford, CT.
The Nature Conservancy. Oriental Bittersweet: Element Stewardship Abstract. In: Wildland Weeds Management & Research Program, Weeds on the Web.
Virginia Native Plant Society. 1995. Invasive Alien Plant Species of Virginia: Oriental Bittersweet (Celastrus orbiculatus Thunb.).

Plant Conservation Alliance, Alien Plant Working Group.

**Management and Control Information**

Return to List




## Species Management and Control Information

# Oriental bittersweet
# *Celastrus orbiculatus* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Oriental bittersweet - http://www.se-eppc.org/manual/bittersweet.html - 2003**

Since Oriental bittersweet produces numerous seeds, extensive seed reserves can become established in the soil within a year or two. Seeds of Oriental bittersweet remain viable for several years and control actions must continue until seed sources are eliminated.

**The Nature Conservancy - Species Management Summary (Element Stewardship Abstract): Oriental Bittersweet - http://tncweeds.ucdavis.edu/esadocs/celaorbi.html - Author: Glenn D. Dreyer - March 3, 1994**

Effective biological and additional effective chemical/mechanical control methods are needed. Education of nursery growers, retailers and the gardening public is also needed to reduce the demand for and the dissemination of the vine and its fruit.

Management Requirements:
Celastrus orbiculatus poses a serious threat to individual plants and plant communities due to its high reproductive rate, long range dispersal, ability to rootsucker, and rapid growth rates. Individual plants can be severely damaged and even killed by the aggressive growth habits of this vine. Tree and shrub stems are weakened and killed by the twining and climbing growth which twists around and eventually constricts solute flow (as shown by Lutz, 1943 for C. scandens). Trees with girdled stems and large amounts of vine biomass in their canopies are more susceptible to damage by wind, snow and ice storms (Siccama, et al. 1976, Langdon 1993). All types of plants, and even entire plant communities, can be over topped and shaded by the vine's rapid vegetative growth. Nearly pure stands of this vine are not uncommon in affected areas. Upland meadows, thickets and young forests, both natural and managed, appear to be most vulnerable to C. orbiculatus dominance.

Langdon (1993) notes that many of the rarest plants in the southeastern U.S. require a natural disturbance regimen of a certain quality and frequency. Because many of these processes have been altered some of these species are now relegated to roadway and utility corridors which provide exactly the sort of habitat most often invaded and dominated by C. orbiculatus. Langdon (1993) also points out that the region's old growth forests such as cove hardwood stands lose 1-2% of their canopy each year which may provide C. orbiculatus with opportunities to invade. Fortunately, it has not yet been found in virgin forests in the Smokies.

Beaches are also open to invasion. In Connecticut, TNC is managing a C. orbiculatus infestation in sand dunes adjacent to a Piping Plover nesting area on Long Island Sound. The managers are concerned that the vines will either spread into actual nesting areas or alter the dynamics of dune formation and erosion (Lapin 1992). In either case, they could interfere with the reproduction of a bird officially listed as a Threatened Species by the State of Connecticut. Very vigorous patches of C. orbiculatus have also been observed growing in pure sand in coastal Rhode Island (Dreyer, pers. obser.).

It is considered of particular concern to forestry programs in some parts of the southern U.S. (McNab and Meeker 1987). The problems throughout the East are most noticeable along roadsides where vegetation is blanketed by bittersweet in a way reminiscent of Kudzu infested areas of the Southeastern U.S.

Given the fact that hybrids with C. scandens are clearly possible, and that C. scandens appears to be less common than in the recent past (Dreyer et al. 1987, Mehrhoff, 1986) the potential for introgressive hybridization, resulting in the loss of C. scandens genetic identity, is possible. Connecticut has recently listed C. scandens as a Species of Special Concern, i.e. one for which more information on distribution and abundance is needed. In Great Smoky Mountains National Park C. scandens is restricted to circumneutral soils and is considered a non-reproducing rare plant (Langdon 1993).

Recovery of natural areas highly infested with C. orbiculatus is unpredictable. Previous natural vegetation structure and function are often severely altered, although remnants of the flora may persist. Removal methods often further disrupt remnants of previous plant communities. A number of workers report that even with complete removal and rootkill of C. orbiculatus, substantial seedling regeneration occurs in following years, probably due to a persistent soil seed bank. Langdon (1993) stresses that individual clones are difficult to kill. For example, one 5m x 5m clone treated with triclopyr in 1986 has produced 50+ sprouts each year since. The sprouts are hand-pulled but often break and resprout later. Fortunately, the six years of work at this site appear to have nearly exhausted the seed bank. In cases where all nearby seed sources cannot be eliminated, however, reinfestation is a continual possibility.

Due to the ease of and apparent interest in cultivating C. orbiculatus in the eastern half of the U.S. and elsewhere, a very large geographic area is potentially threatened. The spread of the vine from coastal States westward should be monitored. In addition, the status of C. scandens populations, especially in east coast States, should be evaluated.

Langdon (1993) strongly suggests that natural areas be scouted for infestations about 2 weeks after the autumn foliage peak. In the Smokies this falls around November 10. By this time other native deciduous plants drop almost all of their leaves while C. orbiculatus leaves turn lemon- to golden-yellow making the plants easy to identify even at a distance or from a vehicle at moderate speeds. Since individual plants are usually exclusively male or female and the seeds may disperse a kilometer or more these types of searches are essential to locate individuals that have recently become established in previously uninfested areas. If carried out conscientiously, such searches will enable managers to implement control programs that contain infestations to particular areas and prevent them from spreading.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Oriental bittersweet - http://www.nps.gov/plants/alien/fact/ceor1.htm - Authors: Carole Bergmann and Jil M. Swearingen -March 2, 1999**

Where hand labor is practical, vines can be pulled out by the roots and removed from the site, preferably before fruiting. If fruits are present, vines should be bagged and disposed of in a landfill, or left in the bags and allowed to bake in the sun long enough to kill the seeds.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Oriental bittersweet - http://www.se-eppc.org/manual/bittersweet.html - 2003**

Cutting: Cut climbing or trailing vines as close to the root collar as possible. This technique is feasible on small populations; as a pretreatment on large impenetrable sites; in areas where herbicide cannot be used; or if labor resources are not sufficient to adequately implement herbicidal control. This treatment will prevent seed production and strangulation of surrounding woody vegetation. Oriental bittersweet will resprout unless cut so frequently that its root stores are exhausted. Treatment should begin early in the growing season and be repeated at two-week intervals until autumn.

Grubbing: This method is appropriate for small initial populations or environmentally sensitive areas where herbicides cannot be used. Using a pulaski or similar digging tool, remove the entire plant, including all roots and runners. Juvenile plants can be hand pulled depending on soil conditions and root development. Any portions of the root system not removed will potentially resprout. All plant parts, including mature fruit, should be bagged and disposed of in a trash dumpster to prevent reestablishment.

**The Nature Conservancy - Species Management Summary (Element Stewardship Abstract): Oriental Bittersweet - http://tncweeds.ucdavis.edu/esadocs/celaorbi.html - Author: Glenn D. Dreyer - March 3, 1994**

MOWING: Regular, weekly mowing will exclude C. orbiculatus. However, less frequent mowing, eg. 2-3 mowings per year, stimulates rootsuckering (Dreyer, pers. obs.).

**Illinois Vegetation Management Guideline - Round-leaved bittersweet (Celastrus orbiculatus) - http://www.inhs.uiuc.edu/chf/outreach/VMG/rlbitter.html - Vol.1, No. 20 - Author: Max Hutchison - February 6, 1990**

RECOMMENDED PRACTICES IN NATURAL COMMUNITIES OF HIGH QUALITY: Where practical, individual vines should be pulled up by the roots and removed from the area by hand. To maintain control, round-leaved bittersweet should be totally eradicated from the surrounding area where possible. Invading individuals should be pulled immediately and removed upon discovery.

## CHEMICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Oriental bittersweet - http://www.nps.gov/plants/alien/fact/ceor1.htm - Authors: Carole Bergmann and Jil M. Swearingen -March 2, 1999**

Certain systemic herbicides, such as glyphosate (e.g., Roundup) or triclopyr (e.g., Garlon), that are taken into the roots and kill the entire plant, have been used successfully in bittersweet management. This method is most effective if the stems are first cut by hand or mowed and herbicide is applied immediately to cut stem tissue. In areas where spring wildflowers or other native plants occur, application of herbicides should be conducted prior to their emergence, delayed until late summer or autumn, after the last killing frost occurs, or carefully targeted. Herbicidal contact with desirable plants should always be avoided. No biological controls are currently known for oriental bittersweet.

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Oriental bittersweet - http://www.se-eppc.org/manual/bittersweet.html - 2003**

Stump Treatment: Use this method in areas where vines are established within or around non-target plants, or where vines have grown into the canopy.

> Glyphosate: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of glyphosate and water to the cross-section of the stem. This procedure is effective at temperatures (as low as 40°F) and may require a subsequent foliar application of glyphosate.
> Triclopyr: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of triclopyr and water to the cross-section of the stem This procedure remains effective at low temperatures (<60°F) as long as the ground is not frozen. A subsequent foliar application may be necessary to control new seedlings.

Foliar Spray Method: Use this method to control large populations. It may be necessary to precede foliar applications with stump treatments to reduce the risk of damaging non-target species.

> Glyphosate: Apply a 2% solution of glyphosate and water plus 0.5% non-ionic surfactant to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves.
> Glyphosate is a non-selective systemic herbicide that may kill non-target partially sprayed plants. Ambient air temperature should be above 65°F.
> Triclopyr: Apply a 2% solution of triclopyr and water to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. The ideal time to spray is after surrounding native vegetation has become dormant (October-November) to avoid affecting non-target species. A 0.5% concentration of a non-ionic surfactant is recommended in order to penetrate leaf cuticle. Ambient air temperature should be above 65°F

**The Nature Conservancy - Species Management Summary (Element Stewardship Abstract): Oriental Bittersweet - http://tncweeds.ucdavis.edu/esadocs/celaorbi.html - Author: Glenn D. Dreyer - March 3, 1994**

A successful control technique was developed by Dreyer (1988) for dense, low patches of C. orbiculatus where herbicide use is appropriate. Vegetation in the entire area is cut to the ground early in the growing season and allowed to resurge. Approximately one month later, foliar applications of an herbicide containing triclopyr (Garlon 4, a triclopyr ester, or Garlon 3A, a triclopyr salt) mixed at 1% to 2% in water and applied by backpack sprayer result in essentially 100% rootkill of C. orbiculatus. No off target damage or root uptake by adjacent plants has been noted in over four years of using this technique. The same study found foliar applications of glyphosate (Roundup, Rodeo) and amitrole (Amitrol, Weedazol) were both ineffective in rootkilling C. orbiculatus.

Another advantage to using triclopyr instead of glyphosate is that it does not kill monocots. Thus grasses, sedges, liliaceous plants, etc., will not be killed and will remain to prevent soils from being completely exposed. These remaining plants often dominate sprayed sites a year after treatment. Triclopyr is also the active ingredient, in relatively dilute form, in the Ortho product Brush B Gone which, unlike Garlon, is not a restricted use chemical.

Hutchison (1992) reports foliar applications of a 2,4 D and triclopyr mixture (Crossbow) to C. orbiculatus will effectively "reduce the population" when applied in mid to late October.

In locations where large vines climb high into trees, cutting and treating the vine stump surface with a triclopyr-containing herbicide is a logical procedure. The vine stems hanging in the trees will decompose and fall within two to three years. Hutchison (1992) recommends cut surface treatment with "100% Roundup" (presumably undiluted with water) applied at the time of the last killing frost, but he included no data concerning the effectiveness of this technique.

**Nonnative Invasive Plants of Southern Forests: a field guide for identification and Control: Bush honeysuckles - Gen. Tech. Rep. SRS-62. Asheville, NC, U.S. Department of Agriculture, Forest Service Research Station - http://www.invasive.org/eastern/srs/OAB.html - Author: James H. Miller - 2003**

Recommended control procedures:

- Thoroughly wet all leaves with one of the following herbicides in water with a surfactant (July to October): Garlon 4, Garlon 3A, or a glyphosate herbicide as a 2-percent solution (8 ounces per 3-gallon mix).
- For stems too tall for foliar sprays, apply Garlon 4 as a 20-percent solution in commercially available basal oil, diesel fuel, or kerosene (2.5 quarts per 3-gallon mix) with a penetrant (check with herbicide distributor) to the lower 16 inches of stems. Or, cut large stems and immediately treat the cut surfaces with one of the following herbicides in water with a surfactant: Garlon 4 or a glyphosate herbicide as a 25-percent solution (32 ounces per 1-gallon mix).

**Illinois Vegetation Management Guideline - Round-leaved bittersweet (Celastrus orbiculatus) - http://www.inhs.uiuc.edu/chf/outreach/VMG/rlbitter.html - Vol.1, No. 20 - Author: Max Hutchison - February 6, 1990**

RECOMMENDED PRACTICES IN NATURAL COMMUNITIES OF HIGH QUALITY: If hand removal is not feasible (e.g. large populations), vines should be cut by hand and cut stems spot-treated with 100% Roundup (a formulation of glyphosphate) just after the last killing frost. A squirt bottle may be used to spot-treat the cut stumps or else herbicide can be wiped on each stump with a sponge applicator. Treatment should be prior to emergence of spring ephemerals. The herbicide applicator should carefully avoid contacting nontarget plants when applying herbicide, because Roundup is nonselective. By law, herbicides only may be applied as per label instructions and by licensed herbicide applicators or operators when working on public properties.

## OTHER CONSIDERATIONS:

**The Nature Conservancy - Species Management Summary (Element Stewardship Abstract): Oriental Bittersweet - http://tncweeds.ucdavis.edu/esadocs/celaorbi.html - Author: Glenn D. Dreyer - March 3, 1994**

The following individuals have experience monitoring and controlling Celastrus orbiculatus:

Glenn Dreyer
The Connecticut College Arboretum
270 Mohegan Avenue
New London, CT 06320

Lise Hanners
Devil's Den Preserve
P.O. Box 1162
Weston, CT 06883
203 226 4991

Keith Langdon
Great Smokey Mt. National Park
107 Park Headquarters Road
Gatlinburg, Tenn. 37738
615 436 1218

W. Henry McNab
USDA Forest Service
SE Forest Experiment Station
Asheville, NC 28806
David Patterson
USDA Ag. Research Service
Botany Departmen
t Duke University
Durham, NC 27706

Paula Piehl
Potomac State College
West Virginia University
Keyser, West Virginia 26726

CONTACTS WITH ADDITIONAL INFORMATION ON CONTROL: In addition to the Connecticut College Arboretum program described above, the following organizations have some type of C. orbiculatus control programs or experience:

Connecticut DOT
Contact: James Stotler
Conn. DOT
24 Wolcott Hill Rd., Drawer A
Wethersfield, CT 06109)

Great Smokey Mountains National Park (Contact is Keith Langdon, see MONITORING PROJECTS section above for address)

Max Hutchison
Cache River Wetlands Projec t
The Nature Conservancy
Route 1, Box 53E
Ullin, Illinois, 62992
618 634 2524

Southeastern Forest Experiment Station (contact is W. Henry McNab, see MONITORING PROJECTS section above for address)

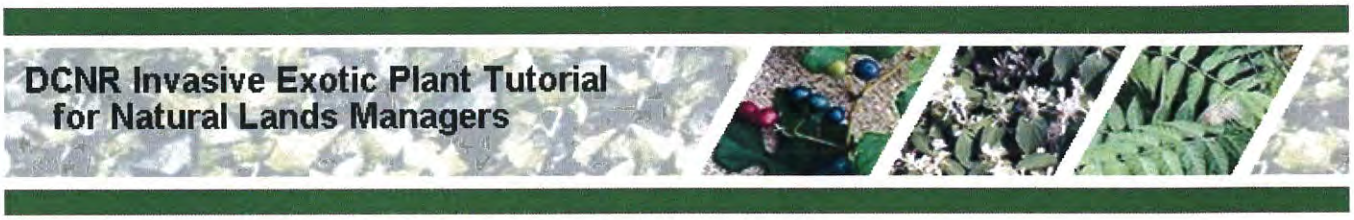
**Illinois Vegetation Management Guideline - Round-leaved bittersweet (Celastrus orbiculatus) - http://www.inhs.uiuc.edu/chf/outreach/VMG/rlbitter.html - Vol.1, No. 20 - Author: Max Hutchison - February 6, 1990**

RECOMMENDED PRACTICES ON BUFFER AND SEVERELY DISTURBED SITES: Same as above in areas where hand labor is available and practical. For large populations in severely degraded sites, foliar spraying with Crossbow (a mixture of 2,4-D and triclopyr), using backpack sprayers, will reduce the popula-tion. Crossbow should be mixed according to label instructions for foliar application. As with Roundup, care should be taken to avoid contacting nontarget plants with herb-icide. Do not spray so heavily that herbicide drips off the target plant. The herbicide should be applied while backing away from the treated area to avoid walking through the wet herbicide. Crossbow is effective when applied in mid-late October.

FAILED OR INEFFECTIVE PRACTICES

- hand control: slow and labor-intensive.
- fire: often not desirable in mesic woodland environments.
- herbicides: should not be used during growing season when spring ephemerals and other native species likely are to be affected.
- no biological controls are known that are feasible in natural areas.

Return to List




## Japanese honeysuckle
## Lonicera japonica Thunb.

**SOURCE:** The following information on this species is taken from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website, Fact Sheets which can be found at http://www.nps.gov/plants/alien/factmain.htm and the Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm. Specific species management and control information is provided elsewhere. Check the website links provided for future updates. The last update for this PCA fact sheet is March 5, 2003.

**NATIVE RANGE**: Japan and Korea
**DESCRIPTION**: Japanese honeysuckle is a perennial vine that climbs by twisting its stems around vertical structures, including limbs and trunks of shrubs and small trees. Stems become woody with maturity, are usually hairy, but sometimes not.




Photo: Chuck Bargeron, The University of Georgia,
www.invasive.org.

Leaves are oblong to oval (reaching 3 inches in length and 2 inches in width), sometimes lobed, have short stalks, occur in pairs along the stem and are hairy. In southern and mid-Atlantic states, Japanese honeysuckle often remains evergreen - its leaves remain attached through the winter. In colder northern climates, the leaves may fall off after exposure to prolonged winter temperatures.







Flowers are tubular, with five fused petals, white to pink, turning yellow with age, very fragrant, and occur in pairs along the stem at leaf junctures. Stems and leaves are sometimes covered with fine, soft hairs. Japanese honeysuckle blooms from late April through July and sometimes into October. Small black fruits are produced in autumn, each containing 2-3 oval to oblong, dark brown seeds about 1/4 inch across.







Photo: Ted Bodner,
Southern Weed Science Society, www.invasive.org

**ECOLOGICAL THREAT**: Exotic bush honeysuckles can rapidly invade and overtake a site, forming a dense shrub layer that crowds and shades out native plant species. They alter habitats by decreasing light availability, by depleting soil moisture and nutrients, and possibly by releasing toxic chemicals that prevent other plant species from growing in the vicinity. Exotic bush honeysuckles may compete with native bush honeysuckles for pollinators, resulting in reduced seed set for native species. In addition, the fruits of exotic bush honeysuckles, while abundant and rich in carbohydrates, do not offer migrating birds the high-fat, nutrient-rich food sources needed for long flights that are supplied by native plant species.




**ECOLOGICAL THREAT**: In North America, Japanese honeysuckle has few natural enemies which allows it to spread widely and out-compete native plant species. Its evergreen to semi-evergreen nature gives it an added advantage over native species in many areas. Shrubs and young trees can be killed by girdling when vines twist tightly around stems and trunks, cutting off the flow of water through the plant. Dense growths of honeysuckle covering vegetation can gradually kill plants by blocking sunlight from reaching their leaves. Vigorous root competition also helps Japanese honeysuckle spread and displace neighboring native vegetation.

**DISTRIBUTION IN THE UNITED STATES**: Japanese honeysuckle occurs across the southern U.S. from California to New England and the Great Lakes region. Escaped populations also occur in Hawaii. Severe winter temperatures and low precipitation may limit its distribution in northern latitudes and in the West, respectively. Click here to see a distribution map.

**HABITAT IN THE UNITED STATES**: A ubiquitous invader, Japanese honeysuckle thrives in a wide variety of habitats including fields, forests, wetlands, barrens, and all types of disturbed lands.

**BACKGROUND**: Japanese honeysuckle was introduced to the U.S. in the early to mid-1800's as an ornamental plant, for erosion control, and for wildlife forage and cover. Its highly fragrant flowers provide a tiny drop of honey-flavored nectar enjoyed by children.

**BIOLOGY & SPREAD**: Growth and spread of Japanese honeysuckle is through vegetative (plant growth) and sexual (seed) means. It produces long vegetative runners that develop roots where stem and leaf junctions (nodes) come in contact with moist soil. Underground stems (rhizomes) help to establish and spread the plant locally. Long distance dispersal is by birds and other wildlife that readily consume the fruits and defecate

the seeds at various distances from the parent plant.

**For more information on Japanese honeysuckle, please contact:**
Ann Rhoads, University of PA, Morris Arboretum (rhoadsaf@pobox.upenn.edu)
Corey Kudrna, National Park Service, Washington, DC (corey_kudrna@nps.gov)
Element Stewardship Abstract, The Nature Conservancy http://tncweeds.ucdavis.edu/esadocs/documnts/lonijap.html
Fire Effects Information System, USDA Forest Service http://www.fs.fed.us/database/feis/plants/vine/lonjap/all.html
Identification and Biology of Non-Native Plants in Florida's Natural Areas, University of Florida http://www.fleppc.org/pdf/Lonicera%20japonica.pdf

Invasive Plant Atlas of New England, University of Connecticut http://webapps.lib.uconn.edu/ipane/browsing.cfm?descriptionid=65
Lisa Jameson, National Park Service, Washington, DC (lisa_jameson@nps.gov)
Nonnative Invasive Plants of Southern Forests - USDA Forest Service http://www.invasive.org/eastern/srs/JH.html
Plant Invaders of Mid-Atlantic Natural Areas, National Park Service and U.S. Fish and Wildlife Service
http://www.invasive.org/eastern/midatlantic/loja.html
Sue Salmons, National Park Service, Rock Creek Park (sue_salmons@nps.gov)
Vikki Nuzzo, Cornell University (vnuzzo@earthlink.net)

SUGGESTED ALTERNATIVE PLANTS: Vines that make good substitutes for Japanese honeysuckle include false jasmine (Gelsemium sempervirens), trumpet honeysuckle (Lonicera sempervirens), trumpet creeper (Campsis radicans), crossvine (Bignonia capreolata), native wisteria (Wisteria frutescens), jackman clematis (Clematis jackmanii), and others. Check with your state native plant society, a reputable native plant nursery, for recommendations for plants that are appropriate for your area and conditions.

**AUTHOR:**
Melissa A. Bravo, National Park Service, Roosevelt-Vanderbilt National Historic Sites, Hyde Park, NY.

**EDITOR:**
Jil M. Swearingen, National Park Service, National Capital Region, Natural Resources and Science, Center for Urban Ecology, Washington, DC.

**REVIEWERS**:
Sylvan Kaufman, Adkins Arboretum, Ridgely, MD.
Corey Kudrna, National Park Service, Washington, DC.
Vikki Nuzzo, Cornell University, Ithaca, NY.

**REFERENCES:**
Barden, L. S. and J. F. Matthews. 1980. Change in abundance of honeysuckle (Lonicera japonica) and other ground flora after prescribed burning of a piedmont pine forest. Castanea 45: 257-260.
Dillenberg L.R., D.F. Whigham, A.H. Teramura, I.N. Forseth. 1993. Effects of below- and aboveground competition from the vines Lonicera japonica and Parthenocissus quinquefolia on the growth of the tree host Liquadambar stryraciflua. Oecologia 93:48-54.
Fernald, M. L. 1989. Grays Manual of Botany. Biosystematics, Floristic and Phylogeny Series. Volume 2. T. R. Dudley, Editor. Dioscorides Press. Portland, OR. 1,632 pp.
Gleason H. A. and A. Cronquist. The Illustrated Companion to Gleason and Cronquist's Manual of Vascular Plants of Northeastern United States and adjacent Canada. New York Botanic Garden, New York, NY. 937 pp.
Kartesz, J. and C. Meacham Synthesis of the North American Flora. Nuzzo, V. Japanese honeysuckle. Element stewardship abstract for Lonicera japonica. The Nature Conservancy. 1815 North Lynn Street, Arlington VA, 22209.
www.tncweeds.ucdavis.edu/esadocs.documnts/lonijap.html. Last updated April 15, 1997. Regehr, D. L. and D. R. Frey. 1988. Selective control of Japanese honeysuckle (Lonicera japonica). Weed Technology 2:139-143.
Rhoads, A. F. and T. H. Block. 2002. The Plants of Pennsylvania, An Illustrated Manual. Morris Arboretum of the University of Pennsylvania. University of Pennsylvania Press, Philadelphia, PA. 1060 pp. Virginia Native Plant Society VA NHP Japanese Honeysuckle Fact Sheet
http://www.vnps.org/invasive/invloni.htm

**Management and Control Information**

Return to List




## Species Management and Control Information

# Japanese honeysuckle
# *Lonicera japonica* Thunb.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese Honeysuckle-**
**http://www.nps.gov/plants/alien/fact/loja1.htm - Author: Melissa Bravo - March 5, 2003**

Several effective methods of control are available for Japanese honeysuckle, including chemical and non-chemical, depending on the extent of the infestation and available time and labor.

**Illinois Vegetation Management Guideline - Japanese honeysuckle (Lonicera japonica)**
**http://www.inhs.uiuc.edu/chf/outreach/VMG/jhnysckl.html - Vol.1, No. 11 - Author: Randy Nyboer - February 6, 1990**

Efforts to control Japanese honeysuckle infestations have included the following methods: mowing, grazing, prescribed burning and herbicides. While grazing and mowing reduces the spread of vegetative stems, prescribed burns or a combination of prescribed burns and herbicide spraying appears to be the best way to eradicate this vine.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese Honeysuckle-**
**http://www.nps.gov/plants/alien/fact/loja1.htm - Author: Melissa Bravo - March 5, 2003**

For small patches, repeated pulling of entire vines and root systems may be effective. Hand pull seedlings and young plants when the soil is moist, holding low on the stem to remove the whole plant along with its roots. Monitor frequently and remove any new plants. Cut and remove twining vines to prevent them from girdling and killing shrubs and other plants. An effective method for removal of patches of honeysuckle covering the ground is to lift up and hold a portion of the vine mass with a rake and have a chain saw operator cut the stems low to the ground. Mowing large patches of honeysuckle may be useful if repeated regularly but is most effective when combined with herbicide application (see below). Mow at twice a year, first in mid-July and again in mid-September. Plants can also be grubbed out using a pulaski or similar digging tool, taking care to remove all roots and runners. Burning removes above ground vegetation but does not kill the underground rhizomes, which will continue to sprout. In certain situations, tethered goats have been used to remove honeysuckle growth, but must be monitored to prevent their escape to the wild where they would become an added ecological threat.

**The Nature Conservancy - Species Management Summary (ESA): Japanese honeysuckle -**
**http://tncweeds.ucdavis.edu/esadocs/lonijapo.html - Authors: Victoria Nuzzo - April 1997**

Mowing, Discing and Pulling: Removing the above-ground portion of Lonicera japonica reduces current-year growth but does not kill the plant, and generally stimulates dense regrowth. Cut material can take root and should therefore be removed from the site (not practical with most infestations).

Mowing is an ineffective control method, stimulating growth and encouraging formation of dense, albeit shorter, mats. Plants mowed in February

formed a dense, 20 cm tall mat within two months, growing from cut stems and rooting from severed runners; by the following November (21 months later) mowed plants were 60 cm high (Stransky 1984). Twice-yearly mowing in Virginia slowed vegetative spread but increased stem density (Williams 1994).

Bush-hogging is an ineffective control, as Lonicera japonica re-invades within one growing season (McLemore 1985).

Discing is apparently an effective control method: McLemore (1985) reported that "control of the honeysuckle was still effective after two years". Discing depth was not indicated. Discing is a highly destructive procedure that destroys native groundlayer species, and may stimulate Lonicera japonica seed bank germination.

Hand-pulling is a time-consuming procedure with limited effectiveness, as the entire plant (roots and shoots) must be removed. Pulling may be a practical method to remove small patches of seedlings.

**The Nature Conservancy - Species Management Summary (ESA): Control Comments on the ESA for Japanese honeysuckle**
**http://tncweeds.ucdavis.edu/esadocs/lonijapo.html - Authors: Marc Imlay - February 2002**

Marc Imlay (ialm@erols.com, 18 February 2002) has a comment on manual removal: The ESA says:

> Hand-pulling is a time-consuming procedure with limited effectiveness, as the entire plant (roots and shoots) must be removed. Pulling may be a practical method to remove small patches of seedlings.

Hand pulling is highly effective over hundreds of acres in our experience. We pull out Japanese honeysuckle by the roots in winter wherever we see it up in the trees, aim the roots upward and tie them in place. The absence of light energy causes the trailing vines to decline precipitously next year. Thus we control 90% of the honeysuckle with 10% of the effort and minimal soil disturbance. We do not pull it out of the trees and watch for native vines (moonseed, trumpet vine, native grape etc.). This method greatly reduces spraying requirements. Use Roundup (glyphosate) or triclopyr in winter or late fall to avoid natives.

## PRESCRIBED FIRE:

**The Nature Conservancy - Species Management Summary (ESA): Japanese honeysuckle -**
**http://tncweeds.ucdavis.edu/esadocs/lonijapo.html - Authors: Victoria Nuzzo - April 1997**

Fire removes above-ground vegetation, and reduces new growth, but does not kill most Lonicera japonica roots, and surviving roots produce new sprouts that return to pre-burn levels of cover within a few years (Oosting and Livingstone 1964). A single spring fire reduced Japanese honeysuckle cover 50% in Illinois (Nyboer 1990). Two sequential fires topkilled Lonicera japonica, reducing crown volume (m3/ha) by 80%, but new growth from root sprouts maintained Lonicera japonica as a dominant groundcover species in North Carolina (Barden and Matthews 1980). In Virginia burning is used to reduce abundance of Lonicera japonica, and inhibit spread for 1-2 growing seasons (Williams 1994). Prescribed burning significantly reduced Lonicera japonica biomass in Tennessee, by 93% when burned in October, and by 59% when burned January - March (Faulkner et al. 1989). Top-killed honeysuckle resprouted in spring (March - April), apparently from roots or runners just below the unburned litter layer. In this situation, follow-up application of 2% glyphosate in spring, 2 - 6 months after burning, appeared to control honeysuckle better on unburned than burned plots, possibly because tall herbaceous vegetation that grew up after the fire on the burned plots intercepted the herbicide before it could reach the shorter honeysuckle resprouts (Faulkner et al. 1989). In Texas, burning in February removed all above ground foliage, but did not kill plants (Stransky 1984). However, burned plants produced fewer and shorter runners than unburned plants, and fire therefore reduced total vegetative growth (Stransky 1984).

Combining fire and herbicides may prove to be more effective than either method by itself if late autumn or winter burns are used to reduce Japanese honeysuckle biomass when most native species are dormant and all resprouts are then treated with a foliar application of glyphosate about a month after they emerge (Johnson, personal communication). Prescribed burns may also be used to help prevent spread of Japanese honeysuckle because seedlings and young plants are most susceptible to fires (Richter, personal communication).

**Illinois Vegetation Management Guideline - Japanese honeysuckle (Lonicera japonica)**
**http://www.inhs.uiuc.edu/chf/outreach/VMG/jhnysckl.html - Vol.1, No. 11 - Author: Randy Nyboer - February 6, 1990**

In fire-adapted communities, spring prescribed burns greatly reduced Japanese honeysuckle coverage and crown volume. Repeated fires reduced honeysuckle by as much as 50% over a single burn. A previously burned population of honeysuckle will recover after several years if fire is excluded during this time. By reducing honeysuckle coverage with fire, refined herbicide treatments may be applied, if considered necessary, using less chemical.

## BIOCONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese Honeysuckle-**
**http://www.nps.gov/plants/alien/fact/loja1.htm - Author: Melissa Bravo - March 5, 2003**

No biological control agents are currently (2003) available for Japanese honeysuckle.

**The Nature Conservancy - Species Management Summary (ESA): Japanese honeysuckle -**
**http://tncweeds.ucdavis.edu/esadocs/lonijapo.html - Authors: Victoria Nuzzo - April 1997**

The only technique that could control Lonicera japonica on a regional scale is biological control, but as of 1997 no formal program had been established. Interestingly, in China, a biocontrol program using Sclerodermus spp. was established to protect Lonicera japonica from the

cerambycid Xylotrechus grayi (Tian et al. 1986). Lonicera japonica is utilized by some insects in its native habitat and the U.S. In Sichuan, China, Lonicera japonica growing near cottonfields is an early spring host for aphids that feed on crops later in the growing season (Li and Wen 1988). In North Carolina, the two-spotted spider mite (Tetranychus urticae), an agricultural pest in corn and peanut fields, overwinters on Lonicera japonica growing on field margins (Margolies and Kennedy 1985). Lonicera japonica is also a suitable host for the cicadellid cotton pest (Empoasca biguttula) in Hunan, China (Chen et al. 1987), and may be a host for tobacco leaf curl virus, which was detected in the horticultural variety Lonicera japonica var. aureo-reticulata (Macintosh et al. 1992). The vine is susceptible to honeysuckle latent virus (Brunt et al. 1980), and to tobacco leaf curl bigeminivirus (TLCV) transmitted by whiteflies (MacIntosh et al. 1992).

## CHEMICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Factsheet - Japanese Honeysuckle- http://www.nps.gov/plants/alien/fact/loja1.htm - Author: Melissa Bravo - March 5, 2003**

In moderate cold climates, Japanese honeysuckle leaves continue to photosynthesize long after most other plants have lost their leaves. This allows for application of herbicides when many native species are dormant. However, for effective control with herbicides, healthy green leaves must be present at application time and temperatures must be sufficient for plant activity. Several systemic herbicides (e.g., glyphosate and triclopyr) move through the plant to the roots when applied to the leaves or stems and have been used effectively on Japanese honeysuckle. Following label guidelines, apply a 2.5% rate of glyphosate (e.g., Rodeo for wetlands; Roundup for uplands) mixed with water and an appropriate surfactant, to foliage from spring through fall. Alternatively, apply a 2% concentration of triclopyr (e.g., Garlon 3A) plus water to foliage, thoroughly wetting the leaves but not to the point of drip-off. A coarse, low-pressure spray should be used. Repeat applications may be needed. Treatment in the fall, when many non-target plants are going dormant, is best. Also, a 25% glyphosate or triclopyr solution mixed with water can be applied to cut stem surfaces any time of year as long as the ground is not frozen.

**The Nature Conservancy - Species Management Summary (ESA): Japanese honeysuckle - http://tncweeds.ucdavis.edu/esadocs/lonijapo.html - Authors: Victoria Nuzzo - April 1997**

The evergreen and semi-evergreen nature of Lonicera japonica allows application of herbicides when many native species are dormant. Timing of application is critical to effectiveness; in general, applying herbicide shortly after the first killing frost, and before the first hard frost (ca. -4.0oC) is most effective. Herbicide effectiveness can be reduced in areas where large stones or fallen logs protect root crowns from soil-active herbicides (Miller 1985) or where overtopping vegetation intercepts foliar herbicides (Faulkner et al. 1989). Many herbicides produce a short-term reduction in foliar coverage, but do not kill the plant and buds left undamaged by the herbicide can produce new growth that often exceeds growth from untreated plants within a year (Prine and Starr 1971). A foliar application of 1.5% glyphosate shortly after the first frost appears to be the most effective treatment. Treated plants should be re-examined at the end of the second growing season, as plants can recover from herbicide application (McLemore 1981).

GLYPHOSATE (brand names include: Roundup, Rodeo, Accord)

- October applications of 0.75% and 1.5% v/v glyphosate killed 99% of treated Lonicera japonica within six months in Delaware, and few plants resprouted within 30 months of treatment (Regehr and Frey 1988). The two application rates were equally effective. The same experiment conducted in December resulted in 68% mortality at the lower concentration, and 86% mortality at the higher concentration, and regrowth from buds was much greater than in plants treated in October. The authors concluded that timing of application was critical; applying glyphosate within 2 days of the first frost resulted in very high mortality. After the first frost, higher concentrations of glyphosate were needed to achieve somewhat lower mortality. Defoliation after glyphosate treatment was very slow; only 5-15% of leaves were gone one month after treatment, although 78-90% of stems were dead.
- A mid-August application of 2.2 kg/ha glyphosate controlled 83% of actively growing Lonicera japonica in North Carolina; control was reduced under drought conditions (Younce and Skroch 1989). Glyphosate (2 lb active ingredient/gal) at 1 to 1.5 gallons/acre controlled "most" Lonicera japonica in Alabama (Miller 1985).
- In Arkansas, a 6.72 kg active ingredient/ha application resulted in 85% control after one growing season, and 80% control after two growing seasons (McLemore 1981). Lower application rates were less effective two years after treatment.
- Effectiveness of glyphosate increased linearly with increasing herbicide concentration (0.48-4.8% w/w), but no concentration gave complete control with one application; repeated treatment with 4.8% glyphosate produced complete shoot necrosis in only 50% of plants (Ahrens and Pill 1985)
- Efficacy of glyphosate was not increased by addition of surfactants (Younce and Skroch 1989, Regehr and Frey 1988).

DICHLORPROP + 2,4-D

- Dichlorprop mixed with 2,4-D at 3.6 grams active ingredient/liter (1.5% v/v) resulted in 94% mortality when applied within two days of the first frost in October, but only 46% mortality when applied in December. Thirty months after treatment, 14% of stems sprayed in October resprouted, and 75% of stems sprayed in December produced new growth (Regehr and Frey 1988).

2,4-D + PICLORAM (brand names include: Tordon)

- Picloram is a restricted use soil-active herbicide that is prohibited in California, as it is relatively persistent and subject to leaching.
- Tordon 101 (4:1 2,4-D amine + picloram, at 1 to 2 gal/acre) "reduced existing honeysuckle to a few surviving crowns" (Miller 1985). Tordon 10K at 50 lb/acre had similar effectiveness (Miller 1985).
- Tordon 101 at 10 gal acre reduced foliage by 72.5% one year after treatment; a second application of Tordon 101 reduced foliage by a total of 90% one year after re treatment (Prine and Starr 1971)
- A foliar spray of Tordon 101 at 2.8-8.4 kg/ha gave 84-94% control in a pine stand (McLemore 1982), similar to control provided by amitrole at 2.24 and 4.48 kg/ha. (McLemore 1982).

TEBUTHIURON (brand names include: Spike)

- Spike 80W (80% tebuthiuron) and Spike 20p (20% tebuthiuron) provided very effective control when applied at 4-5 lbs active ingredient/acre, "resulting in essentially bare plots with yellowing sprigs of vegetation" (Miller 1985).

DICAMBA (brand names include: Banvel, Brushkiller)

- Banvel 720 (2 lb 2,4-D and 1 lb dicamba) was very effective when applied at 4 gal/acre, but had only partial effectiveness at 3 gallons/acre (Miller 1985).
- Lower rates of Dicamba, as in Brushkiller 4-41 and 10-51, resulted in limited or no mortality (Miller 1985). In fact, Lonicera japonica growth was stimulated by application of Brushkiller 10-51 (Miller 1985).

SULFOMETURON (brand names include: Oust)

- A February application of sulfometuron methyl in South Carolina at .25 lb/acre active ingredient, applied when vegetation is less than 30-45 cm high, is recommended for control of Lonicera japonica in loblolly pine stands (Michael 1985).
- In Georgia, Lonicera japonica was not controlled by a late application of Sulfometuron applied at 3 oz/acre (Withrow et al. 1983)
- Lonicera japonica was almost completely killed (99% mortality) by a May application of 2 oz metsulfuron-methyl + 0.25% surfactant in central Georgia (Edwards and Gonzalez 1986)

INEFFECTIVE

- In Illinois, herbicides that are not used by the Department of Conservation due to ineffectiveness or environmental persistence are: picloram; amitrole; aminotriazolel atrazine; dicamba; dicamba + 2,4-D; 2,4-D; DPX 5648; fenac; fenuron; simazine; and triclopyr (brand names for triclopyr include Garlon 3A, Garlon 4 and Brush-B-Gone) (Nyboer 1990).
- Hexazinone at 2.24 and 6.72 kg Active ingredient/ha was ineffective (McLemore 1981), as was application at 1 or 2 lb active ingredient/acre (Michael 1985). Hexazinone pellets at 8 lb active ingredient/acre reduced Lonicera japonica cover from 100% to 25% cover after three years, while a 2 lb/acre rate resulted in a decrease in cover from 100% to 52% over the same time period (Michael 1984).
- Oryzalin is apparently ineffective, as it is recommended for use in controlling weeds that threaten Lonicera japonica planted as a groundcover (Bowman 1983)
- Brushkiller 10-51 at 1.5 gal/acre "encouraged" growth of Lonicera japonica (Miller 1985). Brushkiller 170 resulted in a 45% decrease in foliar cover one year after June treatment (Prine and Starr 1971).
- June application of 2,4-D (4 lb active ingredient/acre at 10 gal/acre) increased foliar growth of Lonicera japonica by 48% one year after treatment (control plants increased by 0.9%) (Prine and Starr 1971).
- June application of Banvel resulted in increased foliar growth one year after treatment (Prine and Starr 1971).
- Triclopyr in both ester and salt formulations (3 and 4lb/gal, respectively) and as an ester combined with 2,4-D (1 and 2lb/gal respectively) failed to control Lonicera japonica one year after treatment (Dreyer 1988). However, in Illinois the latter formulation is reputedly effective (Nyboer 1990).

**Nonnative Invasive Plants of Southern Forests: a field guide for identification and Control: Japanese honeysuckle - Gen. Tech. Rep. SRS-62. Asheville, NC, U.S. Department of Agriculture, Forest Service Research Station - http://www.invasive.org/eastern/srs/JH.html - Author: James H. Miller - 2003**

Recommended control procedures:

- Apply Escort* with a surfactant to foliage June to August-either by broadcast spraying 2 ounces per acre in water (0.6 dry ounces per 3-gallon mix) or by spot spraying 2 to 4 ounces per acre in water (0.6 to 1.2 dry ounces per 3-gallon mix).
- Or, treat foliage with one of the following herbicides in water with a surfactant (July to October or during warm days in early winter) keeping spray away from desirable plants: a glyphosate herbicide as a 2-percent solution (8 ounces per 3-gallon mix) or Garlon 3A or Garlon 4 as a 3- to 5-percent solution (12 to 20 ounces per 3-gallon mix).
- Or, cut large vines just above the soil surface and immediately treat the freshly cut stem with a glyphosate herbicide or Garlon 3A as a 20-percent solution (2.5 quarts per 3-gallon sprayer) in water with a surfactant July to October (safe to surrounding plants).
- Prescribed burning in spring will reduce dense ground mats and sever climbing vines for more effective herbicide treatments to re-sprouting vines.

* Nontarget plants may be killed or injured by root uptake.

**Illinois Vegetation Management Guideline - Japanese honeysuckle (Lonicera japonica)**
**http://www.inhs.uiuc.edu/chf/outreach/VMG/jhnysckl.html - Vol.1, No. 11 - Author: Randy Nyboer - February 6, 1990**

Because Japanese honeysuckle is semi-evergreen, it will continue to photosynthesize after surrounding deciduous vegetation is dormant. This condition allows managers to detect the amount of infestation, and allows for treatment of the infestation with herbicides without damage to the dormant vegetation.
Glyphosate herbicide (tradename Roundup) is the recommended treatment for this honeysuckle. A 1.5-2% solution (2-2.6 oz of Roundup/gallon water) applied as a spray to the foliage will effectively eradicate Japanese honeysuckle. The herbicide should be applied after surrounding vegetation has become dormant in autumn and before a hard freeze (250F). Roundup should be applied carefully by hand sprayer, and spray coverage should be uniform and complete. Do not spray so heavily that herbicide drips off the target species. Retreatment may be necessary for plants that are missed because of dense growth. Although glyphosate is effective when used during the growing season, use at this time is not recommended in natural areas because of the potential harm to nontarget plants. Glyphosate is non-selective, so care should be taken to avoid contacting nontarget species. Nontarget plants will be important in recolonizing the site after Japanese honeysuckle is controlled.
Crossbow, a formulation of triclopyr and 2,4-D, is also a very effective herbicide that controls Japanese honeysuckle. Crossbow should be mixed according to label instructions for foliar application and applied as a foliar spray. It may be applied at dormant periods, like glyphosate, and precautions given above for glyphosate should be followed when using Crossbow. Either herbicide should be applied while backing away from the treated area to avoid walking through the wet herbicide. By law, herbicides only may be applied according to label instructions and by

licensed herbicide applicators or operators when working on public properties.

## OTHER CONSIDERATIONS:

**Illinois Vegetation Management Guideline - Japanese honeysuckle (Lonicera japonica)**
**http://www.inhs.uiuc.edu/chf/outreach/VMG/jhnysckl.html - Vol.1, No. 11 - Author: Randy Nyboer - February 6, 1990**

Maintenance control:
In fire-adapted communities, periodic spring burning should control this species.

FAILED OR INEFFECTIVE PRACTICES:
Mowing limits the length of Japanese honeysuckle vines, but will increase the number of stems produced.
Grazing may have the same effects as mowing, but is less predictable due to uneven treatment given by browsing animals.
Herbicides that have given poor control results or that are more persistent in the environment than other types are picloram, annitrole, aminotriazole, atrazine, dicamba, dicamba & 2,4-D, 2,4-D, DPX 5648, fenac, fenuron, simazine & triclopyr.

Return to List




## Mile-a-Minute Weed
## *Polygonum perfoliatum* L.

**SOURCE**: The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is April 23, 1999.

**NATIVE RANGE:** India to Eastern Asia, China and the Islands from Japan to the Phillipines, including Nepal, Burma, Manchuria, China, Korea, Taiwan and the Malay Peninsula

**DESCRIPTION:** Mile-a-minute weed, also known as Devil's tail tearthumb, is an herbaceous, annual, trailing vine. It has a reddish stem that is armed with downward pointing hooks or barbs which are also present on the underside of the leaf blades. The light green colored leaves are shaped like an equilateral (equal-sided) triangle and alternate along the narrow, delicate stems. Distinctive circular, cup-shaped leafy structures, called ocreas, surround the stem at intervals.




Photo: Jil M. Swearingen, USDI National Park Service, www.forestryimages.org.




Photo: Yun Wu, USDS Forest Service, www.forestryimages.org.

Flower buds, and later flowers and fruits, emerge from within the ocreas. Flowers are small, white and generally inconspicuous. The fruits are attractive, metallic blue and segmented, each segment containing a single glossy, black or reddish-black seed.

**ECOLOGICAL THREAT**: Mile-a-minute weed grows rapidly, scrambling over shrubs and other vegetation, blocking the foliage of covered plants from available light, and reducing their ability to photosynthesize, which stresses and weakens them. If left unchecked, the lack of photosynthesis will kill a plant. Large infestations of mile-a-minute weed eventually reduce native plant species in natural areas. Small populations of extremely rare plants may be eliminated entirely. Because it can smother tree seedlings, mile-a-minute weed has a negative effect on Christmas tree farms, forestry operations on pine plantations and reforestation of natural areas. It has the potential to be a problem to nursery and horticulture crops that are not regularly tilled as a cultivation practice.

Photo: USDA APHIS Archives, USDA APHIS, www.forestryimages.org.




**DISTRIBUTION IN THE UNITED STATES**: Mile-a-minute weed is currently found in Pennsylvania, Maryland, Delaware, West Virginia, New York, Virginia, Ohio and Washington, D.C. These states and the District of Columbia comprise about 20 percent of the estimated possible range for this species. It is considered a temperate species with subtropical tendencies and therefore has the potential to invade those portions of the contiguous United States that have the appropriate climate to provide a minimal 8 week cold vernalization period. A temperature of 10 degrees C or below must be sustained for an 8 week period to stimulate germination.

**HABITAT IN THE UNITED STATES:** Mile-a-minute weed generally colonizes open and disturbed areas, along the edges of woods, wetlands, stream banks, and roadsides, and uncultivated open fields, resulting from both natural and human causes. Natural areas such as stream banks, parks, open space, road shoulders, forest edges and fence lines are all typical areas to find mile-a-minute. It also occurs in environments that are extremely wet with poor soil structure. Available light and soil moisture are both integral to the successful colonization of this species. It will tolerate shade for a part of the day, but needs a good percentage, 63-100% of the available light. The ability of mile-a-minute to attach to other plants with its recurved barbs and climb over the plants to reach an area of high light intensity is a key to its survival. It can survive in areas with relatively low soil moisture, but demonstrates a preference for high soil moisture.

**BACKGROUND**: The first records of mile-a-minute in North America are from Portland, Oregon (1890) and Beltsville, Maryland (1937). Both of these sites were eliminated or did not establish permanent populations of the species. However, the introduction of mile-a-minute in the late 1930's to a nursery site in York County, Pennsylvania did produce a successful population of this plant. It is speculated that the seed was spread with Rhododendron stock. The owner of the nursery was interested in the plant and allowed it to reproduce; unfortunately, subsequent efforts to eradicate it were not successful. The distribution of mile-a-minute has radiated from the York County site into neighboring states. In the past 55 years, the range for this plant in the United States has extended as far as 300 miles in several directions from the York County, Pennsylvania site (Mountain, 1995) and (Okay 1997).

**BIOLOGY & SPREAD:** Mile-a-minute weed is primarily a self-pollinating plant (supported by its inconspicuous, closed flowers and lack of a detectable scent), with occasional outcrossing. Fruits and viable seeds are produced without assistance from pollinators. Vegetative propagation from roots has not been successful for this plant. It is a very tender annual, withering with a slight frost, and reproduces successfully until the first frost. Mile-a-minute is a prolific seeder, producing many seeds on a single plant over a long season, from June until October in Virginia, and a slightly shorter season in more northern geographic areas. Birds are probably the primary long-distance dispersal agents of mile-a-minute weed. Transport of seeds short distances by native ant species has been observed. This activity is probably encouraged by the presence of a tiny white food body (elaiosome) on the tip of the seed that may be attractive to the ants. These seed-carrying ants may play an important role in the survival and germination of the seeds of mile-a-minute weed. Local bird populations are important for dispersal under utility lines, bird feeders, fence lines and other perching locations. Other animals observed eating mile-a-minute weed fruits are chipmunks, squirrel and deer. Water is also an important mode of dispersal for mile-a-minute weed. Its fruits can remain buoyant for 7-9 days, an important advantage for dispersing seed long distances in stream and river environments. The long vines frequently hang over waterways, allowing fruits that detach to be carried away in the water current. During storm events the potential spread of this plant is greatly increased throughout watersheds.

**AUTHORS:** Judith A. Gerlach Okay, Virginia Department of Forestry, Fairfax, VA.

**EDITOR:** Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on mile-a-minute weed, please contact:**
Cornell University, Biological Control of Weeds-Mile-a-Minute Weed, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
Invasive Plant Atlas of New England, http://invasives.eeb.uconn.edu/ipane/
Jil Swearingen, U.S. National Park Service, National Capital Region, Washington DC; 202-342-1443, ex. 218.
Judy Okay, Virginia Department of Forestry, 12055 Government Center Parkway, Fairfax, VA 22035; 703-324-1480.
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild Fact Sheets, http://www.nps.gov/plants/alien/fact/pocu1.htm.
Plant Invaders of Mid-Atlantic Natural Areas, http://www.nps.gov/plants/alien/pubs/midatlantic/
Sue Salmons, U.S. National Park Service, Rock Creek Park, Washington, DC; 202-426-6834, ex. 33.
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program Fact Sheet-Mile-a-Minute Weed; http://www.dcr.virginia.gov/dnh/invlist.htm

**REFERENCES:**
Gruber, Alfred. 1995. Unpublished. Polygonums - Mile-a-minute weed and Mile-a-minute vine. Mile-a-minute (Polygonum perfoliatum) Conference. York, Pennsylvania, July.
Hill, Rovert J., G. Springer, and L.B. Forer. 1981. Mile-a-minute, Polygonum perfoliatum L. (Polygonaceae), a New Potential Orchard and Nursery

Weed. Regulatory Horticulture 7(1). Pennsylvania Dept. of Agriculture.
McCormick, L.H. 1995. Mile-a-minute Control in Reforestation. Mile-a-minute (Polygonum perfoliatum) Conference. York, Pennsylvania, July.
Moul, E.T. 1948. A dangerous weedy Polygonum in Pennsylvania. Rhodora 50:64-66.
Mountain, W. L. 1995. Mile-a-minute - History Distribution and Habitat. Mile-a-minute (Polygonum perfoliatum) Conference. York, Pennsylvania, July.
Okay, J.A. Gerlach. 1997. Polygonum perfoliatum: A Study of Biological and Ecological Features Leading to the Formation of a Management Policy. Ph.D. Dissertation. George Mason University Fairfax, VA.
Park, C.W. 1986. Taxonomy of Polygonum section Echinocaulon. Ph.D. Thesis, Cornell, University.
Swearingen, Jil. 1991. Seed dispersal by ants. The Maryland Natural Naturalist, vol. 35, no.1-4. pp. 31-39.

**Plant Conservation Alliance, Alien Plant Working Group.**

**Management and Control Information**

Return to List




## Species Management and Control Information

# Mile-a-Minute Weed
# *Polygonum perfoliatum* L.

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## MANUAL AND MECHANICAL CONTROL:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Mile-a-Minute Weed - http://www.nps.gov/plants/alien/fact/pope1.htm - Author: Judith A. Gerlach - April 23, 1999.**

A variety of control measures, including physical, mechanical, cultural and chemical, can be used for management of mile-a-minute weed. Hand pulling of seedlings is best done before the recurved barbs on the stem and leaves harden. But may be done afterwards with the help of thick gloves. Removal of vines by hand may be conducted throughout the summer, if tough gloves and protective clothing (coveralls) are worn to avoid the skin shredding ability of the recurved hooks. The delicate vines can be reeled in fairly easily and balled up in piles that can be left to dehydrate for several days before disposal. The site must be rechecked at frequent intervals, and removal of new plants continued until the seed germination period is complete, roughly early April until early July in the middle Atlantic states. Repeated mowing or trimming of mile-a-minute plants will prevent the plants from flowering and thus reduce or eliminate fruit and seed production.

Cultural methods can be utilized to discourage the introduction of mile-a-minute to an area. It is important to maintain vegetative community stability and to avoid creating gaps or openings in existing vegetation. Maintaining broad vegetative buffers along streams and forest edges will help to shade out and prevent establishment of mile-a-minute weed. This will also help to reduce the dispersal of fruits by water.

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Alien Plant Species of Virginia - Species Fact Sheets - Mile-a-Minute Weed - http://www.dcr.virginia.gov/dnh/invlist.htm**

Because mile-a-minute is an annual (propagating only from seeds) with a shallow root system, this invasive is best removed from lightly infested areas by digging the plants up (with strong gloves to protect hands from the spines) and disposing of them before they go to seed. Seed set begins in early August and terminates at first frost. Removal of the plants is also best ac- complished before the plant becomes excessively viney. Removal of brush, leaves and woodpiles which may create thick litter is also effective in controlling the spread of the plant.

## BIOCONTROL:

**USDA Forest Service - Biological Control of Invasive Plants in the Eastern United States, http://www.invasive.org/eastern/biocontrol/26MileAMinute.html - Publication FHTET-2002-04 - Authors: R. Van Driesche, et al. - 2002**

No releases of agents have yet been made against Mile-a-Minute Weed.

## CHEMICAL CONTROL*:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Mile-a-Minute Weed - http://www.nps.gov/plants/alien/fact/pope1.htm - Author: Judith A. Gerlach - April 23, 1999.**

An herbicidal soap, of which there are several brands available at most garden centers, will help to burn back foliage of mile-a-minute. Because these products do not have the systemic (ie., travels to the roots) ability of herbicides like glyphosate, they will need to be reapplied all season long to any regrowth. Glyphosate (e.g., Roundup for upland areas and Rodeo for wetland applications), applied at a low rate will probably be effective in killing mile-a-minute weed. However, because this plant is not currently listed on the product labels for Roundup or Rodeo, treatments with these products is permissable only with prior approval of the State Department of Agriculture where the application will take place [FIFRA 1997, Section 2(z)(ee)].

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Alien Plant Species of Virginia - Species Fact Sheets - Mile-a-Minute Weed - http://www.dcr.virginia.gov/dnh/invlist.htm**

Herbicides may be used as an alternative in heavily infested areas. Spot applications of biodegradable glyphosphate herbicides are recommended before mile-a-minute goes to seed in early August. As glyphosphate is a non-selective herbicide which affects all green vegetation, it should be used sparingly so as not to contact desirable vegetation which may be growing with the mile-a-minute. Professionals should be consulted to determine the best method of control in patches that are heavily infested with this invasive species.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




# Kudzu
## *Pueraria montana* var. *lobata* (Willd.) Maesen & S. Almeida

**SOURCE**: The following information on this species is taken, in part, from the Plant Conservation Alliance, Alien Plant Working Group, Weeds Gone Wild website at http://www.nps.gov/plants/alien/factmain.htm. Modifications include additional pictures (from www.invasives.org) and resource information. Check the website links for future updates. The last update for this fact sheet is May 5, 2005.

**NATIVE RANGE**: Asia
**DESCRIPTION**: Kudzu ia a climbing, semi-woody, perennial vine in the pea family. Deciduous leaves are alternate and compound, with three broad leaflets up to 4 inches across. Leaflets may be entire or deeply 2-3 lobed with hairy margins. Individual flowers, about 1/2 inch long, are purple, highly fragrant and borne in long hanging clusters. Flowering occurs in late summer and is soon followed by production of brown, hairy, flattened, seed pods, each of which contains three to ten hard seeds.










Photos: Ted Bodner, Southern Weed Science Society, www.forestryimages.org.

**ECOLOGICAL THREAT:** Kudzu kills or degrades other plants by smothering them under a solid blanket of leaves, by girdling woody stems and tree trunks, and by breaking branches or uprooting entire trees and shrubs through the sheer force of its weight. Once established, Kudzu plants grow rapidly, extending as much as 60 feet per season at a rate of about one foot per day. This vigorous vine may extend 32-100 feet in length, with stems 1/2 - 4 inches in diameter. Kudzu roots are fleshy, with massive tap roots 7 inches or more in diameter, 6 feet or more in length, and weighing as much as 400 pounds. As many as thirty vines may grow from a single root crown.




Photo: Kerry Britton, USDA Forest Service, www.forestryimages.org.

**DISTRIBUTION IN THE UNITED STATES:** Kudzu is common throughout most of the southeastern U.S. and has been found as far north as

Pennsylvania

**HABITAT IN THE UNITED STATES:** Kudzu grows well under a wide range of conditions and in most soil types. Preferred habitats are forest edges, abandoned fields, roadsides, and disturbed areas, where sunlight is abundant. Kudzu grows best where winters are mild, summer temperatures are above 80 degrees Farenheit, and annual rainfall is 40 inches or more.

**BIOLOGY & SPREAD:** The spread of kudzu in the U.S. is currently limited to vegetative expansion by runners and rhizomes and by vines that root at the nodes to form new plants. Kudzu also spreads somewhat through seeds, which are contained in pods, and which mature in the fall. However, only one or two viable seeds are produced per cluster of pods and these hard-coated seeds may not germinate for several years.

**BACKGROUND**: Kudzu was introduced into the U.S. in 1876 at the Philadelphia Centennial Exposition, where it was promoted as a forage crop and an ornamental plant. From 1935 to the mid-1950s, farmers in the south were encouraged to plant kudzu to reduce soil erosion, and Franklin D. Roosevelt's Civilian Conservation Corps planted it widely for many years. Kudzu was recognized as a pest weed by the U.S. Department of Agriculture and, in 1953, was removed from its list of permissible cover plants.

**SUGGESTED ALTERNATIVE PLANTS:** Native vines such as trumpet creeper (*Campsis radicans*), pipevine (*Aristolochia macrophylla*), passionflower (*Passiflora lutea*), trumpet honeysuckle (*Lonicera sempervirens*), and native bittersweet (*Celastrus scandens*) have attractive flowers and fruits, provide food for wildlife and make excellent substitutes for kudzu. These plants should be used in landscaping and for land restoration where they are known to occur as natives.

**AUTHORS:** Carole Bergmann, Montgomery County Department of Park and Planning, Silver Spring, MD. Jil M. Swearingen, U.S. National Park Service, Washington, DC.

**For more information on kudzu, contact:**
Cornell University, Biological Control of Weeds--Kudzu, http://www.invasiveplants.net/invasiveplants/biologicalcontrol/default.asp
James H. Miller, USDA Forest Service, DeVall Drive, Auburn University, AL 36849 Miller@forestry.Auburn.edu
Mid-Atlantic Exotic Pest Plant Council, Inc., http://www.ma-eppc.org
National Invasive Species Information Center, http://www.invasivespeciesinfo.gov
Southeast Exotic Pest Plant Council; Invasive Plant Manual, http://www.se-eppc.org/manual/
Stratford Kay, Aquatic and Noncropland Weed Management, Crop Science Department, Box 7620, North Carolina State University, Raleigh, NC 27695, (919) 515-5645 Stratford_Kay@ncsu.edu
The Bugwood Network, MA-EPPC Plant List, http://www.invasive.org/maweeds.cfm
USDA Forest Service, Nonnative Invasive Plants of Southern Forests, http://www.invasive.org/eastern/srs/
USDA Forest Service, Northeastern Area, Invasive Plants: Weeds of the Week, http://www.na.fs.fed.us/fhp/invasive_plants/weeds/
USDA - NRCS PLANTS Database, http://plants.usda.gov/
Virginia Natural Heritage Program Fact Sheet--Kudzu; http://www.dcr.virginia.gov/dnh/invlist.htm
Virginia Tech Weed Identification Guide, http://www.ppws.vt.edu/weedindex.htm

**REFERENCES:**
McKnight, B.N., ed. 1993. Biological Pollution. Indiana Academy of Sciences, Indianapolis, IN. 261 pp.
Miller, J.H. and B. Edwards. 1982. Kudzu: Where did it come from? And how can we stop it? Southern Journal of Applied Forestry. Pp. 165-169.
Randall, J.M. and J. Marinelli. 1996. Invasive Plants:Weeds of the Global Garden. Brooklyn Botanic Garden Club, Inc. Handbook No. 149. 111 pp.
Virginia Native Plant Society. 1995. Invasive alien plant species of Virginia: kudzu [Pueraria lobata (Willd.) Ohwi]. Virginia Department of Conservation and Recreation, Richmond, Virginia.

**Plant Conservation Alliance, Alien Plant Working Group.**

**Management and Control Information**

Return to List




## Species Management and Control Information

# Kudzu
# *Pueraria montana* var. *lobata* (Willd.) Maesen & S. Almeida

**Source:** The following information is taken from a number of sources and those sources are identified at the beginning of the referenced information. The intent of this resource is to provide the user with as much of the information that exists for management and control of this species as is practical. It is important to note that new and improved methods are added regularly which will require you to visit the websites provided for updates on this information. Bibliographies and resources referenced by these sources are not included here, but are included at the websites from which this information was extracted.

**READ THIS FIRST!**

Before administering any of the following management and control options, it is imperative that you are familiar with the background information provided under the General Management and Control Section.

For additional, and potentially more current, information on management and control of this species, use the Mid-Atlantic Exotic Pest Plant Council (MA-EPPC) listserve or any of the other listserves identified in the Resource - Listserve section of this tutorial. You will find directions for subscribing to the list serve there. The MA-EPPC listserve has an archives feature that saves past discussions (beginning in 1999) about specific species control. These messages are at: http://groups.yahoo.com/group/ma-eppc/messages.

## GENERAL MANAGEMENT CONSIDERATIONS:

**Plant Conservation Alliance - Alien Plant Working Group - Weeds Gone Wild - Fact sheet - Kudzu- http://www.nps.gov/plants/alien/fact/pulo1.htm - Authors: Carole Bergmann and Jil M. Swearingen - May 5, 2005.**

For successful long-term control of kudzu, the extensive root system must be destroyed. Any remaining root crowns can lead to re-infestation of an area. Mechanical methods involve cutting vines just above ground level and destroying all cut material. Close mowing every month for two growing seasons or repeated cultivation may be effective. Cut kudzu can be fed to livestock, burned or enclosed in plastic bags and sent to a landfill. If conducted in the spring, cutting must be repeated as regrowth appears to exhaust the plant's stored carbohydrate reserves. Late season cutting should be followed up with immediate application of a systemic herbicide (e.g., glyphosate) to cut stems, to encourage transport of the herbicide into the root system. Repeated applications of several soil-active herbicides have been used effectively on large infestations in forestry situations. Efforts are being organized by the U.S. Forest Service to begin a search for biological control agents for kudzu.

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Plant Fact Sheets - http://www.dcr.virginia.gov/dnh/invlist.htm.**

Control of well established kudzu stands can take up to 10 years. Persistent eradication of all roots is the key to the control of this pest, keeping in mind that a single kudzu patch may extend past landowner boundaries. The most effective method of control will depend on several factors; size of the infested area, proximity to sensitive species or other desirable vegetation, and accessibility of the patch. Small patches of kudzu that are not well established (usually ones less than ten years old) can be eliminated by persistent weeding, mowing, or grazing during the growing season over a period of three to four years. Unfortunately, with root systems that can be up to 12 feet deep, eradication by direct root removal is not practical.

## MANUAL AND MECHANICAL CONTROL:

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Kudzu - http://www.se-eppc.org/manual/kudzu.html -2003**

Grubbing: Using a pulaski or similar digging tool, remove the entire plant, including the taproot. Removed vegetation should be destroyed by burning or bagging. Because many roots exceed 1.8 m, eradication by this method is very difficult and should be considered primarily for small initial incursions.

Cutting: Vines and runners are chopped just above the ground level, and the pieces destroyed. Early in the season, cutting is repeated at two-week intervals, to weaken the crown and prevent resumption of photosynthesis. Later in the season, when the stored energy in the taproot has been reduced, the interval between cuttings can be extended. Cutting does not typically kill roots and should only be used to control the spread of kudzu.

## PRESCRIBED FIRE:

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Plant Fact Sheets - http://www.dcr.virginia.gov/dnh/invlist.htm.**

In some areas, prescribed burning may be used as a follow-up treatment after herbicide application. Although it should be carefully and professionally handled, this two step process is effective in clearing out leaf litter and speeding recolonization of an area by desirable native plant species.

## BIOCONTROL:

**USDA Forest Service - Biological Control of Invasive Plants in the Eastern United States, http://www.invasive.org/eastern/biocontrol/25Kudzu.html - Publication FHTET-2002-04 - Authors: R. Van Driesche, et al. - 2002**

Preliminary host range testing with soybean and peanut were begun in the summer of 2000. As far as possible, initial host testing will be conducted in China, where quarantine facilities are not required for these native insects. In the later screening stages, extensive testing of American plants and crops will be conducted in U.S. quarantine facilities to ensure host specificity before any insect can be released.

## CHEMICAL CONTROL*:

**Southeast Exotic Pest Plant Council - Invasive Plant Manual - Kudzu - http://www.se-eppc.org/manual/kudzu.html -2003**

Cut Stump Method: Use this method in areas where vines are established within or around non-target plants or where vines have grown into the canopy.

- Glyphosate: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of glyphosate and water to the cross-section of the stem. This procedure is effective at temperatures as low as 40°F, and may require a subsequent foliar application of glyphosate.
- Triclopyr: Cut the stem 5 cm (2 in) above ground level. Immediately apply a 25% solution of triclopyr and water to the cross-section of the stem. This procedure remains effective at low temperatures (<60°F) as long as the ground is not frozen. A subsequent foliar application may be necessary to control new seedlings.

Foliar Spray Method: Use this method to control large populations. It may be necessary to precede foliar applications with stump treatments to reduce the risk of damaging non-target species. After the stems and leaves have been brought under control (i.e., all above ground portions of the plants have been effectively treated) further treatment should follow the Root Crown Method.

- Glyphosate: Apply a 2% concentration of glyphosate and water plus a 0.5% non-ionic surfactant to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. Glyphosate is a non-selective systemic herbicide that may kill non-target partially-sprayed plants. Ambient air temperature should be above 65°F.
- Triclopyr: Apply a 2% concentration of triclopyr and water to thoroughly wet all foliage. Do not apply so heavily that herbicide will drip off leaves. A 0.5% concentration of a non-ionic surfactant is recommended in order to penetrate leaf cuticle. Ambient air temperature should be above 65°F.

Root Crown Method: Follow the young or resprouting stem of the plant to the root. Dig and cut into the root crown using a pulaski or similar tool. Apply a 50% glyphosate solution or 50% triclopyr solution to the main root crown and any below ground runners.

**Virginia Department of Conservation and Recreation and the Virginia Native Plant Society - Invasive Plant Fact Sheets - http://www.dcr.virginia.gov/dnh/invlist.htm.**

Long-term treatment of heavily infested sites usually requires the application of herbicides over a period of up to five years to inhibit the growth of new shoots. Biodegradable glyphosphate herbicides are recommended for control of kudzu in natural areas. These herbicides break down rapidly into harmless components when exposed to soil. Because glyphosphate is a systemic, non-selective herbicide that affects all green vegetation, treatments should be carefully timed and applied by trained applicators. The best time for application of these herbicides is at the end of the growing season when the plants are actively transporting nutrients from leaves and stems to root systems. When applied at the proper time, herbicides are transported to the roots where they are able to kill the entire plant.

**USDA Forest Service - Nonnative Invasive Plants of Southern Forests: a field guide for identification and control - http://www.invasive.org/eastern/srs/ - Gen. Tech. Rep. SRS-62. Asheville, NC - Author: James H. Miller - 2003.**

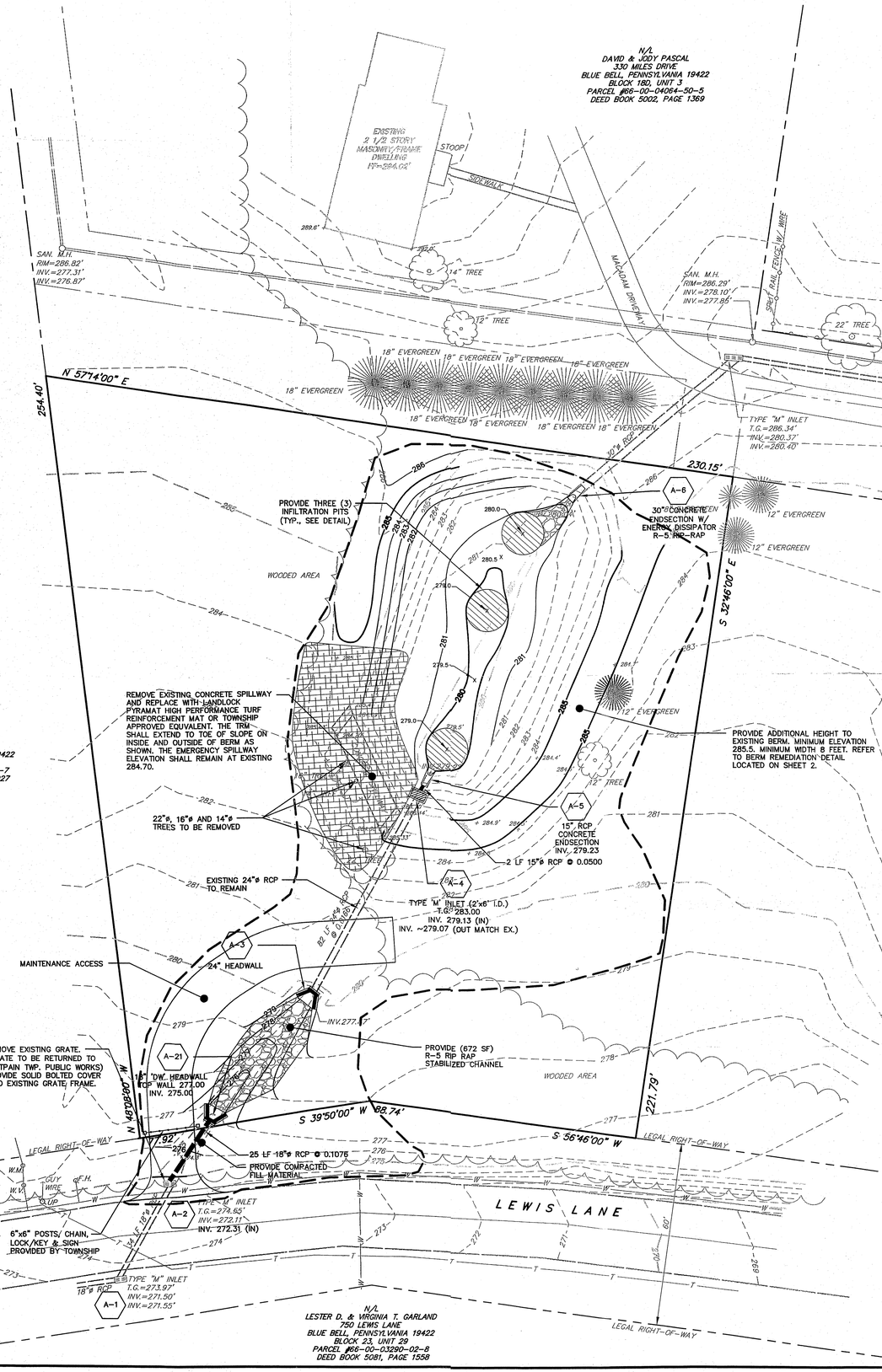
Recommended control procedures:

- Thoroughly wet all leaves (until runoff) with one of the following herbicides in water with a surfactant: July to October for successive years when regrowth appears - Tordon 101* as a 3-percent solution (12 ounces per 3-gallon mix) or Tordon K* as a 2-percent solution (8 ounces per 3 - gallon mix), either by broadcast or spot spray - spraying climbing vines as high as possible. July to September for successive years - Escort** at 3 to 4 ounces per acre in water (0.8 to 1.2 dry ounces per 3-gallon mix) - or when safety to surrounding vegetation is desired, Transline*** as a 0.5 percent solution in water (2 ounces per 3-gallon mix); spray climbing vines as high as possible or cut vines that are not controlled after herbicide treatment.
- For partial control, repeatedly apply Garlon 4 or a glyphosate herbicide as a 2-percent solution in water (8 ounces per 3-gallon mix) with a surfactant during the growing season. Cut large vines and immediately apply these herbicides to the cut surfaces. Or, apply Garlon 4 as a 20-percent solution in commercially available basal oil, diesel fuel, or kerosene (2.5 quarts per 3-gallon mix) with a penetrant (check with herbicide distributor) to large vines as a basal spray (January to April), which controls vines less than 2 inches in diameter.

*Nontarget plants may be killed or injured by root uptake. When using Tordon herbicides, rainfall must occur within 6 days after application for needed soil activation. Tordon herbicides are Restricted Use Pesticides.
**Nontarget plants may be killed or injured by root uptake.
*** Transline controls a narrow spectrum of plant species.

* Mention of pesticide products in this document does not constitute endorsement of any particular material.

Return to List




## GENERAL REQUIREMENTS

1. DO NOT MOW OR USE WEED WHACKERS NEAR TREES & SHRUBS.

2. DO NOT MOW TO LESS THAN RECOMMENDED MOWING HEIGHT.

3. INSPECT ALL SEEDED AREAS FOR BARE SPOTS (GREATER THAN 1 SQUARE FOOT) IN LATE SUMMER. RESEED BARE SPOTS WITH APPROPRIATE SEED MIXES DURING THE FALL SEEDING WINDOW (AUGUST 15 – SEPTEMBER 30) OR SPRING SEEDING WINDOW (APRIL 15 – JUNE 15).

4. REPLENISH MULCH AROUND TREES AND SHRUBS EACH SPRING TO SUPPRESS WEEDS. REMOVE WEEDS BY HAND AS NEEDED. VIGOROUS WEED CONTROL IS NEEDED FOR THE FIRST THREE YEARS OR UNTIL TREES ARE TALL ENOUGH TO SHADE OUT COMPETING WEEDS.

5. MONITOR FOR DEER DAMAGE DURING ESTABLISHMENT YEARS, USE REPELLENT AS NEEDED.

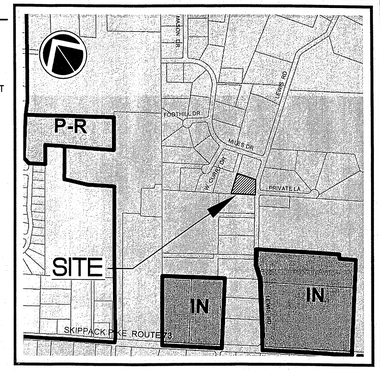
6. MONITOR AND REMOVE INVASIVE SPECIES IN ALL AREAS. IT IS RECOMMENDED THAT AN INVASIVE SPECIES EXPERT BE CONTRACTED FOR ANNUAL OR BIANNUAL VISITS TO ASSIST IN IDENTIFICATION AND ELIMINATION OF ANY LOCAL INFESTATIONS OF BIENNIAL OR PERENNIAL WEEDS OR INVASIVE WOODY SPECIES.

7. KEEP PHOTO RECORDS AND WRITTEN LOSS AS A RECORD OF SUCCESSES AND FAILURES OF PLANTING AND MANAGEMENT TECHNIQUES. TAKE PHOTOS WITHIN THE FIRST MONTH AFTER INSTALLATION ('BEFORE') AND ONCE EACH SEASON (FOUR TIMES ANNUALLY). TAKE PHOTOS FROM THE SAME LOCATION SO THAT CHANGES CAN BE COMPARED.

8. DURING FIRST THREE YEARS OF BASIN RETROFIT ESTABLISHMENT COORDINATE MAINTENANCE AND TRAINING OF TOWNSHIP STAFF WITH MAINTENANCE CONTRACTOR.

9. INLET AND OUTLET STRUCTURES SHOULD BE INSPECTED AND CLEANED IF NECESSARY A MINIMUM OF 2 TIMES PER YEAR OR AFTER LARGE STORM EVENTS (GREATER THAN 3 INCHES IN 24 HOURS). IF IT IS FOUND THAT THE CLOGGING OF THE INLET AND OUTLET STRUCTURES IS OCCURRING FREQUENTLY, IT MAY BE NECESSARY TO INSPECT/CLEAN STRUCTURES MORE OFTEN.

10. INLETS SHOULD BE INSPECTED FOR SIGNS OF EROSION AND IF PRESENT, SHALL BE REGRADED AND REPAIRED. TURF REINFORCEMENT MATTING OR EROSION CONTROL BLANKET SHOULD BE INSTALLED IN THE AFFECTED AREA AND THE AREA SHOULD BE REVEGETATED IMMEDIATELY.




SITE LOCATION MAP 1"= 800'

## FIRST YEAR MAINTENANCE

TURF GRASS:

1. MOW TURF AREAS AS SOON AS TOP GROWTH IS TALL ENOUGH TO CUT. MOW GRASS TO 3 INCHES HIGH. REPEAT MOWING TO MAINTAIN SPECIFIED HEIGHT WITHOUT CUTTING MORE THAN 40 PERCENT OF GRASS HEIGHT. DO NOT MOW WHEN GRASS IS WET.

MEADOW AREAS:

1. MOW MEADOW AREAS TO A HEIGHT OF 5 INCHES WHEN GROWTH REACHES 10 INCHES IN HEIGHT.

2. MOW WITH A FLAIL–TYPE MOWER EVERY 10 WEEKS THROUGH THE GROWING SEASON. REMOVE OR FINELY CHOP AND REDISTRIBUTE MOWINGS TO PREVENT CUT WEEDS FROM SMOTHERING NATIVE GRASS SEEDLINGS. THE USE OF A FLAIL–TYPE MOWER IS RECOMMENDED.

TREE/SHRUB GROVES:

1. WEED AS NECESSARY TO REMOVE INVASIVE SPECIES.

2. MULCH WITH COMPOSTED SHREDDED LEAF LITTER TO A DEPTH OF 3 INCHES.

3. WATER OR SUPPLEMENT NATURAL RAINFALL TO PROVIDE A MINIMUM RATE OF 1 INCH PER WEEK.

## SECOND YEAR AND BEYOND MAINT.

TURF GRASS:

1. MOW TURF AREAS AS SOON AS TOP GROWTH IS TALL ENOUGH TO CUT. MOW TO 3 INCHES HIGH. REPEAT MOWING TO MAINTAIN SPECIFIED HEIGHT WITHOUT CUTTING MORE THAN 40 PERCENT OF GRASS HEIGHT. DO NOT MOW WHEN GRASS IS WET.

MEADOW AREAS:

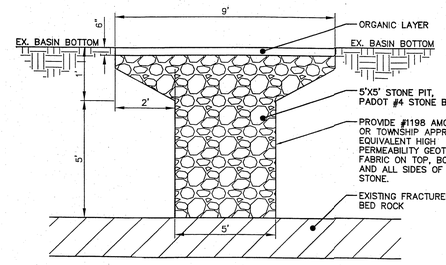
1. MOW MEADOW AREAS TO A HEIGHT OF 8 INCHES IN MID SPRING BEFORE WARM SEASON GRASSES EMERGE, BUT WHEN COOL SEASON WEEDS ARE ACTIVELY GROWING. IF WET MEADOW AREAS ARE TOO WET TO MOW IN SPRING, MOW IN LATE FALL. REMOVE OR FINELY CHOP AND REDISTRIBUTE MOWINGS.

2. INSPECT 1–2 TIMES ANNUALLY FOR INVASIVE SPECIES. IF BIENNIAL WEEDS SUCH AS SWEET CLOVER, WILD PARSNIP, QUEENS ANN'S LACE, OR BURDOCK APPEAR, THEY SHOULD BE MOWED OR WEED WHACKED TO A HEIGHT OF 12 INCHES OR LESS WHEN IN BLOOM. DO NOT ALLOW BIENNIAL WEEDS TO GO TO SEED.

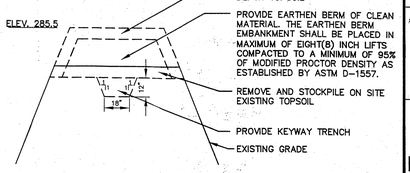
TREE/SHRUB GROVES:

1. WEED AS NECESSARY TO REMOVE INVASIVE SPECIES.

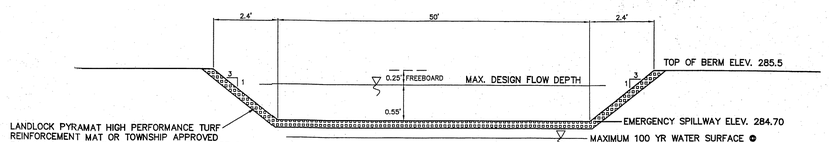
2. MULCH WITH COMPOSTED SHREDDED LEAF LITTER TO A DEPTH OF 3 INCHES IN NECESSARY TO SUPPRESS WEED GROWTH OR ADD TO NATURAL LEAF LITTER.




INFILTRATION PIT DETAIL




BERM REMEDIATION DETAIL




LANDLOCK PYRAMAT HIGH PERFORMANCE TURF REINFORCEMENT MAT OR TOWNSHIP APPROVED EQUIVALENT LINING SHALL EXTEND TO ELEV. 283 ON THE OUTSIDE OF THE BASIN BERM AND TO ELEV. 281 ON THE INSIDE OF THE BASIN BERM..

BASIN 64 EMERGENCY SPILLWAY


Land Concepts
215-646-2988
593 Skippack Pike, Suite 300
Blue Bell, Pennsylvania 19422
www.landconceptsgroup.com
IN CONJUCTION WITH:
HIBBELN ENGINEERING COMPANY, LLC
CONSULTING ENGINEERS
593 Skippack Pike • Suite 300 • Blue Bell, PA 19422
LAND SURVEYING:
TRACY LAND SERVICES, INC.
21 EAST LINCOLN AVENUE
HATFIELD, PA 19440
(215) 855-5500


# VALENTINE ESTATES BASIN (BASIN #64) REMEDIATION

PREPARED FOR
WHITPAIN TOWNSHIP
SITE SITUATE IN
WHITPAIN TOWNSHIP
MONTGOMERY COUNTY, PENNSYLVANIA

| NO. | REVISIONS | PLAN ORIGINATION DATE |
|---|---|---|
| 1 | PER MCCD COMMENTS | FEBRUARY 1, 2010 |
| | | OCTOBER 2, 2009 |


ALL INSTRUMENTS PREPARED BY HIBBELN ENGINEERING COMPANY, L.L.C. ARE REPRESENTATIVE OF SERVICES RENDERED OR TO BE RENDERED BY HIBBELN ENGINEERING COMPANY, L.L.C. IN CONNECTION WITH THE PROJECT FOR WHICH THEY WERE PREPARED AND ARE NOT TO BE USED FOR ANY OTHER PROJECT OR EXTENSION OF THIS PROJECT EXCEPT BY AGREEMENT OR WITH THE APPROVAL OF HIBBELN ENGINEERING COMPANY, L.L.C. ALL REUSE, CHANGES, LOSS, OR EXTENSION IS WITHOUT LIABILITY OF HIBBELN ENGINEERING COMPANY, L.L.C.



Land Concepts
593 Skippack Pike, Suite 300
Blue Bell, Pennsylvania 19422
www.landconceptsgroup.com


## MAINTENANCE, OPERATIONS AND IMPROVEMENTS PLAN

VALENTINE ESTATES BASIN (BASIN #64) REMEDIATION
SITE SITUATE IN
WHITPAIN TOWNSHIP
MONTGOMERY COUNTY, PENNSYLVANIA

| | |
|---|---|
| PROJECT MANAGER: DSC/AJH | DRAFTED BY: MLW/JBS |
| PROJECT NUMBER: 543 | DRAWING FILE: #543-64_SHT02 |
| PLAN SCALE: 1"= 20' | PLAN SHEET NUMBER 2 OF 6 |

0' 10' 20' 40'
GRAPHIC SCALE