



PennEast Pipeline Company, LLC

PENNEAST PIPELINE PROJECT

Pennsylvania Invasive Species Management Plan

August 2019

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ACRONYM LIST

APHIS	Animal and Plant Health Inspection Service
BMP	best management practice
CFR	Code of Federal Regulations
EI	Environmental Inspector
ISMP	Invasive Species Management Plan
NWA	Noxious Weed Act
NWCL	Noxious Weed Control Law
PADCNR	Pennsylvania Department of Conservation of Natural Resources
PCS	Pennsylvania Consolidated Statutes
PDA	Pennsylvania Department of Agriculture
PennEast	PennEast Pipeline Company, LLC
Project	PennEast Pipeline Project
PS	Pennsylvania Statutes
ROW	right-of-way
SLF	spotted lanternfly
TCD	Thousand Cankers Disease
USC	United States Code
USDA	U.S. Department of Agriculture

1.0 INTRODUCTION

PennEast Pipeline Company, LLC (PennEast) has prepared this Invasive Species Management Plan (ISMP) for the Pennsylvania portion of the proposed PennEast Pipeline Project (Project). The Project's pipeline facilities, located in Pennsylvania and New Jersey, are identified by segment and would include the PennEast mainline route 36-inch diameter pipeline, the 4-inch diameter Blue Mountain Lateral, the 24-inch Hellertown Lateral, the 12-inch diameter Gilbert Lateral, and the 36-inch diameter Lambertville Lateral. The PennEast mainline route would be an approximately 115-mile long new pipeline starting in Luzerne County, Pennsylvania, and extending to Mercer County, New Jersey. Approximately 77 miles of the mainline route pipeline is located in Pennsylvania, crossing portions of Luzerne, Carbon, Monroe, Northampton, and Bucks Counties. See Figure 1: Project Overview Map for visual reference.

PennEast has prepared this ISMP to avoid and minimize the spread of invasive species during construction of the Project in Pennsylvania. The movement of vehicles, equipment, and personnel, and the transport of materials and/or construction debris to and from areas that are inhabited by invasive species could result in the unintentional spread of these species during construction. Additionally, recently disturbed areas may provide an opportunity for the colonization and spread of invasive species. This ISMP will serve as a guidance document to minimize the potential spread of invasive species during construction. This guidance document is specific to BMPs that should be implemented within the workspace required to construct the pipeline and has not been prepared to address offsite mitigation sites.

2.0 EXISTING CONDITIONS

The proposed Project crosses multiple land use types in Pennsylvania, from human-altered landscapes such as residential, agricultural, commercial/industrial, transportation corridors (roadways, railroad), and utility transmission corridors, to relatively undisturbed natural landscapes such as forested uplands, open land, forested wetlands, non-forested wetlands, and watercourses. Human-altered landscapes often create suitable conditions for establishment of the quick-germinating, fast-growing, nutrient-poor soil-loving species that typically characterize invasive plant species. Although the Project area remains predominantly rural, it has had a long history of settlement since colonial times, with agriculture and timber harvesting as the predominant industries having influenced land development. Consequently, even the relatively undisturbed lands can consist of long abandoned agricultural fields and second- to third-growth woodland, which may have sufficiently altered the natural vegetation community, allowing for establishment of invasive plant species. PennEast has identified a variety of invasive plant, insect, and aquatic species which are known to occur within Pennsylvania counties crossed by the Project, which are listed in the sections below.

2.1 INVASIVE AND NOXIOUS PLANT SPECIES

In Pennsylvania, invasive plants are defined as non-native species that grow aggressively and displace native vegetation. Noxious weeds are a subset of invasive plants that a state or federal agency identifies as being particularly detrimental to public health or natural and economic resources. Such plants are monitored through the federal noxious weed program by the U.S. Department of Agriculture, Animal and Plant Health Inspection Service (APHIS). Federal authority for listing and regulating noxious plant species

comes from the Plant Protection Act of 2000 (7 USC 104), and Noxious Weed Control and Eradication Act of 2004 (7 USC 7701).

The Pennsylvania Department of Agriculture (PDA) also maintains a state-specific list of noxious weeds. In November 2017, Pennsylvania's Noxious Weed Act (NWA) was signed into law, replacing the Noxious Weed Control Law (NWCL), which had been in place since 1982. The NWCL listed 13 species as noxious; under the NWA, new species were listed and noxious plants were separated into three classes (A, B, or C) based on their ability to be managed or eradicated. Class A noxious weeds are geographically limited, and intended to be eradicated. Class B noxious weeds are widely distributed and likely cannot be eradicated, but can be controlled. Class C noxious weeds include any species found on the Federal Noxious Weed List which are not yet established in Pennsylvania and not listed under Class A or B. Refer to Appendix A for a complete list of Pennsylvania's noxious weeds, and Appendix B for identification sheets for Class A and B noxious plants.

To determine which invasive species are likely to be encountered during construction and restoration activities, PennEast referred to the invasive plant species listed as occurring within each Pennsylvania County crossed by the Project in the iMap Invasives web application. Refer to Table 3-1 below for a complete list of invasive plants documented in Pennsylvania Counties crossed by the project, including six (6) Pennsylvania Class B Noxious Weeds (denoted with a †). Plant species listed as noxious weeds by the PDA under the NWA are the focus of the proposed prevention and control measures found in Section 3.1.

Table 2-1 Invasive Plants Documented in Project Counties (PA)

Species Name	Common Name	Recorded Observation by County
<i>Acer platanoides</i>	Norway Maple	Bucks, Monroe, Luzerne
<i>Acorus calamus</i>	Sweetflag	Bucks, Northampton, Carbon
<i>Aegopodium podagraria</i>	Bishop's Goutweed	Northampton
<i>Ailanthus altissima</i>	Tree-of-Heaven	Bucks, Northampton, Luzerne
<i>Alliaria petiolata</i>	Garlic Mustard	Bucks, Northampton, Monroe, Luzerne
<i>Artemisia vulgaris</i>	Mugwort	Bucks, Northampton, Monroe
<i>Berberis thunbergii</i>	Japanese Barberry	Bucks, Northampton, Monroe, Carbon, Luzerne
<i>Berberis vulgaris</i>	European Barberry	Northampton
<i>Celastrus orbiculata</i>	Oriental Bittersweet	Bucks, Monroe
<i>Centaurea biebersteinii</i> / <i>Centaurea stoebe micranthos</i>	Spotted Starthistle/Knapweed	Luzerne
<i>Chelidonium majus</i>	Greater Celandine	Northampton, Monroe
<i>Cirsium arvense</i> †	Canada Thistle	Northampton
<i>Cirsium vulgare</i> †	Bull Thistle	Northampton, Luzerne
<i>Conium maculatum</i> †	Poison-hemlock	Bucks
<i>Datura stramonium</i>	Jimsonweed	Northampton
<i>Dipsacus fullonum</i>	Wild Teasel	Bucks
<i>Elaeagnus umbellata</i>	Autumn Olive	Bucks, Northampton, Monroe, Luzerne
<i>Euphorbia cyparissias</i>	Cypress Spurge	Northampton
<i>Frangula alnus</i>	Glossy False Buckthorn	Northampton
<i>Hemerocallis fulva</i>	Orange Daylily	Bucks
<i>Hesperis matronalis</i>	Dame's Rocket	Bucks, Northampton, Monroe, Luzerne
<i>Holcus lanatus</i>	Common Velvetgrass	Northampton

Species Name	Common Name	Recorded Observation by County
<i>Humulus japonicus</i>	Japanese Hop	Bucks, Northampton
<i>Hypericum perforatum</i>	Common St. John's-wort	Northampton
<i>Iris pseudacorus</i>	Yellow Iris	Bucks, Northampton
<i>Ligustrum obtusifolium</i>	Border Privet	Bucks, Northampton, Monroe
<i>Ligustrum vulgare</i>	European Privet	Bucks, Northampton, Monroe
<i>Lonicera japonica</i>	Japanese Honeysuckle	Bucks, Northampton, Luzerne
<i>Lonicera maackii</i>	Amur Honeysuckle	Northampton, Luzerne
<i>Lonicera morrowii</i>	Morrow's Honeysuckle	Bucks, Northampton, Luzerne, Monroe
<i>Lysimachia nummularia</i>	Creeping Jenny	Bucks, Northampton, Monroe
<i>Lythrum salicaria</i> †	Purple Loosestrife	Bucks, Northampton, Monroe, Luzerne
<i>Microstegium vimineum</i>	Japanese Stiltgrass	Bucks, Northampton, Monroe, Carbon, Luzerne
<i>Morus alba</i>	White Mulberry	Bucks, Northampton, Monroe
<i>Myosotis scorpioides</i>	True Forget-me-not	Northampton
<i>Myriophyllum spicatum</i>	Eurasian Water-milfoil	Bucks, Northampton, Luzerne
<i>Nasturtium officinale</i>	Watercress	Bucks, Northampton, Monroe, Carbon, Luzerne
<i>Ornithogalum umbellatum</i>	Common Star-of-Bethlehem	Bucks
<i>Persicaria hydropiper</i>	Marshpepper Knotweed; smartweed	Bucks, Monroe
<i>Phalaris arundinacea</i>	Reed Canary Grass	Bucks, Northampton, Monroe, Luzerne
<i>Phragmites australis</i>	Common Reed	Bucks, Luzerne
<i>Poa trivialis</i>	Scribner's Bluegrass	Bucks, Northampton, Monroe
<i>Polygonum cuspidatum</i> / <i>Fallopia japonica</i>	Japanese Knotweed	Bucks, Northampton, Monroe, Luzerne
<i>Polygonum perfoliatum</i> / <i>Persicaria perfoliata</i> †	Mile-a-minute-weed	Bucks, Monroe, Luzerne

Species Name	Common Name	Recorded Observation by County
<i>Potamogeton crispus</i>	Curly-leaf Pondweed	Bucks, Northampton, Monroe, Carbon, Luzerne
<i>Pyrus calleryana</i>	Callery Pear	Bucks
<i>Rorippa sylvestris</i>	Creeping Yellowcress	Bucks, Northampton
<i>Rosa multiflora</i> [†]	Multiflora Rose	Bucks, Northampton, Monroe, Luzerne
<i>Rubus phoenicolasius</i>	Wineberry	Bucks, Northampton, Monroe
<i>Salix alba</i>	White Willow	Northampton
<i>Securigera varia</i>	Common Crown-vetch	Bucks, Luzerne
<i>Solanum dulcamara</i>	Climbing Nightshade	Bucks, Northampton
<i>Sonchus arvensis uliginosis</i>	Field Sowthistle	Bucks
<i>Tussilago farfara</i>	Colt's-foot	Monroe, Northampton
<i>Typha angustifolia</i> , <i>Typha x glauca</i>	Narrowleaf Cattail, Hybrid Cattail	Bucks, Northampton, Monroe, Luzerne
<i>Veronica anagallis-aquatica</i>	Water Speedwell	Northampton
<i>Vinca minor</i>	Lesser Periwinkle	Northampton, Luzerne

Source: iMapInvasives (iMapInvasives.natureserve.org), accessed 1/11/2019.

[†]Pennsylvania Class B Noxious Weed

2.2 INVASIVE INSECT SPECIES

Review of the Pennsylvania Department of Conservation of Natural Resources (PADCNR)'s website indicates that several harmful invasive insect species are known to occur within the geographic area of the proposed Project, including:

- Emerald ash borer (*Agrilus planipennis*);
- Hemlock woolly adelgid (*Adelges tsugae*);
- Walnut twig beetle (*Pityophthorus juglandis*)/Thousand Cankers Disease (TCD);
- Gypsy moth (*Lymantria dispar*); and
- Spotted lanternfly (*Lycorma delicatula*).

PennEast's proposed measures to prevent or control the transport of invasive insect species can be found in Section 3.2.

2.3 INVASIVE AQUATIC SPECIES

According to the iMap Invasives web application, several invasive aquatic species have been documented within the Pennsylvania counties impacted by the Project. These species include:

- Rusty crayfish (*Orconectes rusticus*)
- Goldfish (*Carassius auratus*)
- Chinese Mysterysnail (*Cipangopaludina [Bellamya] chinensis*)
- Asiatic Clam (*Corbicula fluminea*)
- Common carp (*Cyprinus carpio*)
- Zebra mussel (*Dreissena polymorpha*)

PennEast's proposed measures to prevent or control the transport of invasive aquatic species can be found in Section 3.3.

3.0 INVASIVE SPECIES MANAGEMENT

The following best management practices (BMPs) will be employed to assist in minimizing the potential transport of invasive species from areas where they may currently occur along the Project route in

Pennsylvania and New Jersey. PennEast will implement the BMPs detailed in this ISMP during all phases of construction. Compliance or non-compliance with these requirements will be recorded in the field and details of the activities will be captured by the Environmental Inspector (EI) during their daily inspection activities.

3.1 MEASURES TO PREVENT OR CONTROL THE TRANSPORT OF NOXIOUS PLANT SPECIES

PennEast will perform the following measures to minimize the potential transport of noxious plant species:

- Prior to construction, PennEast will provide training to educate contractor(s) and subcontractor(s) with respect to the site-specific protocols for controlling transport of invasive plant species within or outside of the Project workspace limits.
- EIs will be employed, by spread, during construction to monitor and provide oversight and implementation of this ISMP.
- Sediment and erosion control devices will be installed across the construction right-of-way (ROW) on slopes upgradient of wetlands and watercourses to prevent soil from migrating offsite during construction. These measures also will help to prevent the dispersion of seeds and root masses from invasive plant species into wetlands, watercourses and drainage ways.
- Equipment used within wetlands will be tracked, often operating on top of construction mats. These measures will minimize the amount of soil disturbance within wetlands in which invasive species might colonize.
- Where feasible, full ROW topsoil segregation will occur in locations densely populated with invasive vegetation. Use of this BMP measure is anticipated to eliminate equipment travel over locations containing invasive seeds, roots or other viable plant materials as topsoil will be stripped, segregated and stockpiled outside of travel ways within the approved limits of the construction ROW.
- To the extent practicable, the contractor(s) and subcontractor(s) will avoid the movement of soils, gravel, rock, and other fill materials infested with invasive plants to locations that are relatively free of invasive plants.
- Contractor(s) and subcontractor(s) will use weed-free mulch, gravel, and fill.
- Construction equipment and vehicle tires will be inspected and cleaned of remnant soils, vegetation, and debris before proceeding to a new construction spread, township, or county as directed by the EI.
- Cleaning procedures will occur at designated locations on equipment mats via the use of shovels, brooms, or other hand tools and/or compressed air or power blowers to clean equipment of dirt, seeds, roots, or other viable plant materials.
- Soil collected at the cleaning stations shall be disposed of in the following manner:
 - Buried onsite within the pipeline trench;
 - Returned back to the point of origin along travel portions of the access road or ROW for site grading; or
 - Collected and transported offsite to a State approved disposal facility;

- After final grading, the restored ROW will be seeded with a seed mix that is free of noxious species to restore herbaceous cover over the disturbed ROW and help prevent establishment of invasive species.
- EIs will be employed during all phases of construction to monitor, provide oversight and enforce implementation of the identified BMPs.

3.2 MEASURES TO PREVENT OR CONTROL THE TRANSPORT OF INVASIVE INSECT SPECIES

Prior to construction, PennEast will provide training to educate contractor(s) and subcontractor(s) with respect to identifying the species listed in Section 2.2, as well as the Asian longhorn beetle (*Anoplophora glabripennis*), which has not yet been identified within the proposed Project area. If any of these insects are found during the course of the Project, they will be reported to the PDA and USDA (U.S. Department of Agriculture). When conducting clearing operations, PennEast's contractor will adhere to the PDA Plant Pest Act regulations (3 P.S. Section 258.1), as well as the USDA federal regulations pertaining to the treatment and transport of materials within quarantine locations (7 Code of Federal Regulations [CFR] Part 301). PennEast will provide contractors with training on the quarantine areas and transport regulations relevant to Project construction during the pre-construction meeting and will provide training refreshers and new staff training as needed throughout the life of the Project.

3.2.1 Spotted Lanternfly Control Measures

The spotted lanternfly (SLF) is an invasive pest that was first detected in Pennsylvania in September 2014. Since 2014, the insect has spread rapidly throughout southeastern Pennsylvania. Long-distance spread is facilitated by people who move infested material or items containing egg masses. This species threatens agriculture including grape, tree-fruit, hardwood, and nursery industries.

The PDA has issued a quarantine for counties where spotted lanternfly has been confirmed. As of August 2019, the quarantine county list included Carbon, Monroe, Northampton, and Bucks counties where the Project is sited. An SLF permit is required for businesses and organizations located or working within the quarantine areas, which move vehicles, products or other conveyances within or out of the quarantine zone. On May 1, 2019, the PDA began working with the Pennsylvania State Police to begin performing compliance and enforcement checks, enforcing SLF permits.

A 2-hour training course must be completed to acquire an SLF permit. After successful completion of the training course, that individual is considered a trainer and is required to supply training to all other employees. Employees should be knowledgeable about what to look for and how to safeguard against moving SLF on products, vehicles, and other conveyances.

In addition to possessing a permit, an SLF inspection checklist must be completed before moving a vehicle, equipment, or materials within or out of the SLF quarantine area. A SLF Checklist for Vehicle Inspection is provided in Appendix E. If any life stages of the SLF are observed, the employee must remove and devitalize the SLF, place it in a sealed bag, and dispose of the bag in the garbage. During an enforcement check, a completed inspection checklist must be presented in addition to the SLF permit. Upon arrival to

the destination, the permit must be hung on the rear-view mirror to confirm that the vehicle was inspected for any life stages of the SLF.

Recordkeeping of training, inspection of vehicles, and control measures taken (such as vehicle washes, destruction of living lanternfly, etc.) must be maintained for a minimum of two years.

3.3 MEASURES TO PREVENT OR CONTROL THE TRANSPORT OF INVASIVE AQUATIC SPECIES

3.3.1 Hydrostatic Pressure Testing

Following pipeline installation, PennEast will hydrostatically pressure test the new pipeline system in accordance with the requirements outlined in CFR Part 192 – Transportation of Natural and Other Gas by Pipeline: Minimum Federal Safety Standards. Prior to testing, PennEast will obtain applicable water withdrawal and discharge permits.

3.3.1.1 Disposal of Hydrostatic Test Water

Water used for hydrostatic testing will be discharged against a splash plate or other approved energy dissipating device to aerate, slow, and disperse the flow within a filtration structure such as a hay bale corral or barrier lined with geotextile fabric. Dewatering will occur in well-vegetated upland areas within the same source watershed as the withdrawal and at a rate of discharge that is appropriate for site conditions to prevent flooding and erosion. The discharge of hydrostatic test water over vegetated upland locations is anticipated to aid in the prevention of aquatic invasive species.

3.3.1.2 Protective Measures

Additional protective measures that will be implemented during hydrostatic testing operations include:

- During the filling of the pipeline, a screened floating water intake structure will be used at each water withdrawal site to prevent the accidental uptake of native or potentially invasive species.
- Piping and withdrawal units will be outfitted with a backflow prevention device to prevent surface water returns during pumping operations.
- The water withdrawal intake apparatus as well as any support equipment used to facilitate its installation will be cleaned and dried before being used at each water withdrawal location.
- All equipment will be inspected prior to onsite operation and will be required to be visibly free of soil, debris or plant material, and fully drained and dry with no residual water before placing the equipment in use. Any visible debris or residual plant material observed would be cleaned as required.

3.3.2 Watercourse Crossings

The procedures for all watercourse crossings associated with the Project will be conducted in accordance with the BMPs outlined in PennEast's permit applications for water obstruction and encroachment as well

as for erosion & sediment control. To minimize the potential for the transport of invasive aquatic species, PennEast will provide training for construction personnel and EIs regarding BMPs that will minimize the potential transport of aquatic invasive species including:

- Restricting equipment and materials to only those necessary for watercourse construction;
- Inspecting materials used to construct the pipeline across watercourses to confirm they are visibly free of vegetation, mud, fish and other aquatic animals prior to entering and exiting a watercourse; and
- Drying equipment and construction materials before entering new watercourses.

3.4 MONITORING FOR INVASIVE PLANT SPECIES

Post-construction monitoring of invasive plant species will be conducted for a period of five (5) years in wetlands and two (2) years in uplands, concurrent with wetland and upland restoration success monitoring. Annual monitoring reports detailing the success of restoration will identify invasive plant species' locations and approximate densities, which will be used to determine if species-specific management measures are necessary. Relative density of invasive species found on the ROW will be compared to the off-ROW densities, and if on-ROW density is significantly greater than the adjacent off-ROW plant community, PennEast will develop a plan to address the invasive species which may include:

- Mechanical removal;
- Biological treatments; or
- If approved by the landowner or land managing agency, and in accordance with regulatory requirements, the use of spot treatment by herbicides.

4.0 SUMMARY/CONCLUSIONS

PennEast will conduct invasive species management within the Project workspace areas in Pennsylvania in a manner that is consistent with the objective of the Project. The proposed management activities outlined within this Plan are expected to help prevent the inadvertent spread of existing populations of invasive species and will promote the establishment of native plant populations.

5.0 REFERENCES

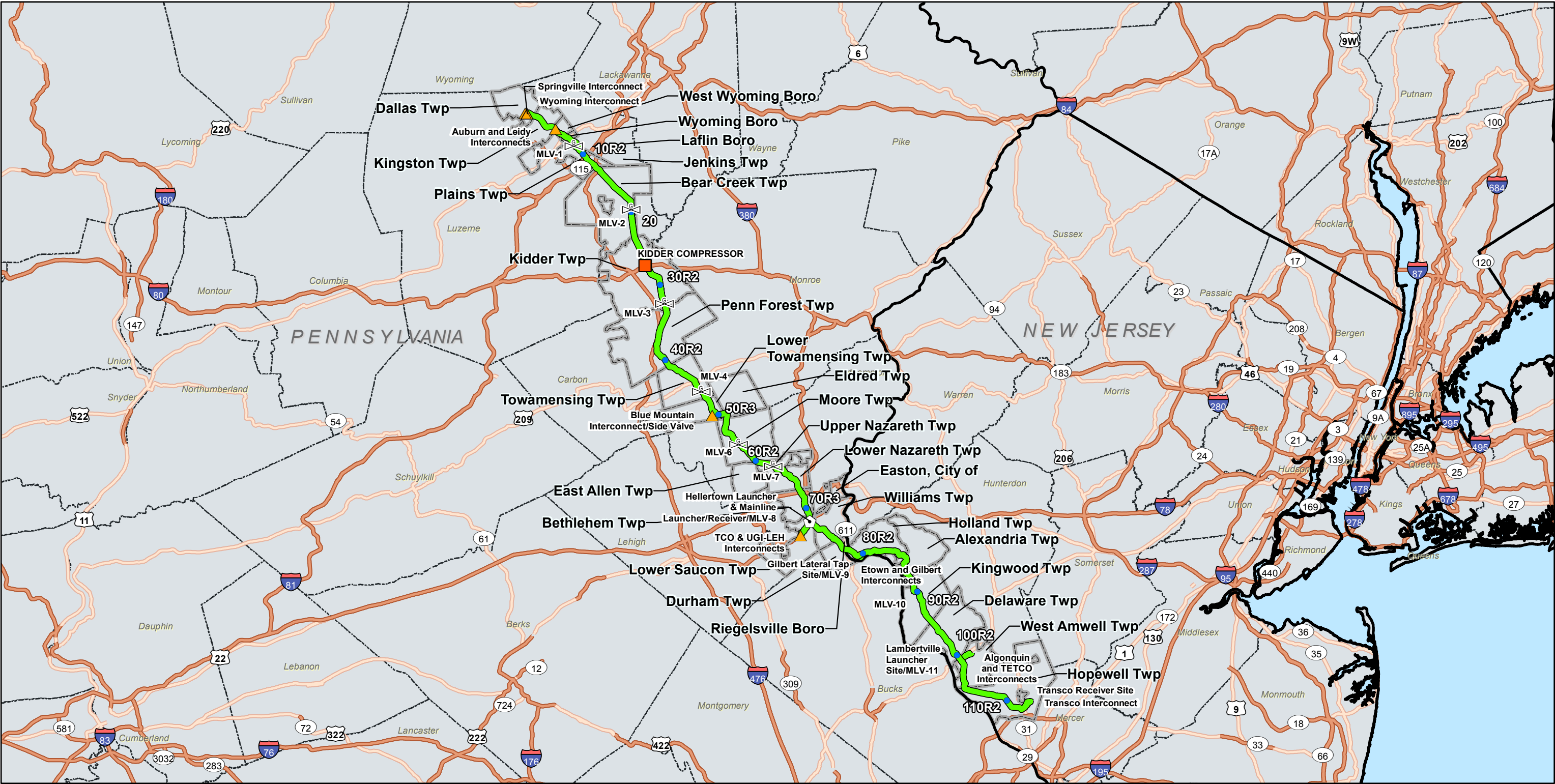
- 7 Code of Federal Regulations (CFR) Part 301. USDA federal regulations pertaining to the treatment and transport of materials within quarantine locations.
- 49 CFR Part 192. Transportation of Natural and Other Gas by Pipeline: Minimum Federal Safety Standards.
- 3 Pennsylvania Statutes (PS) §255. Noxious Weed Control Law.
- 3 PS §258. Plant Pest Act.
- 3 Pennsylvania Consolidated Statutes (PCS) 15. Pennsylvania's NWA.
- 7 United States Code (USC) § 104. 2000. Plant Protection Act.

7 USC §7701. 2004. Noxious Weed Control and Eradication Act.

iMap Invasives. Invasive plant, insect, and aquatic invasives query for Bucks, Northampton, Monroe, Carbon, and Luzerne County, Pennsylvania. iMapInvasives.natureserve.org. Accessed 11 January 2019.

PADCNR. 2017. Forest Insects and Disease. <http://www.dcnr.state.pa.us/forestry/insectsdisease/index.htm>. Accessed January 2018.

Figures



Legend

Mile Post

Launcher/Receiver Site

Interconnect

Mainline Block Valve

Compressor Station

Proposed Route

State Boundary

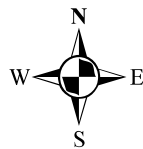
Intersected Municipality Boundary

County Boundary

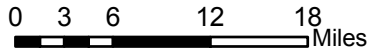
Reference:
esri Streets



PennEast Pipeline Company, LLC



NAD 1983 UTM Zone 18N
Projection: Transverse Mercator
Linear Unit: Meter (1.0)



Key Map Not Drawn to Scale



TITLE:

Figure 1
PennEast Pipeline Project
Project Overview Map

LOC: Luzerne County, Pennsylvania to Mercer County, New Jersey			REV.:
CKD. BY: BH	ENG.:	Date: 10/18/2018	W.O.:
DRN. BY: CDW	SCALE: See Graphic Scale	DWG. NO.:	SHEET: 1

Appendix A
Pennsylvania Noxious Weed List

Pennsylvania Noxious Weed Species

Class A Noxious Weeds

Giant Hogweed	<i>Heracleum mantegazzianum</i>
Goatsrue	<i>Galega officinalis</i>
Kudzu	<i>Pueraria lobata</i>
Palmer amaranth	<i>Amaranthus palmeri</i>
Waterhemp	<i>Amaranthus rudis</i>
Tall waterhemp	<i>Amaranthus tuberculatus</i>
Animated oat	<i>Avena sterilis</i>
Dodder	<i>Cuscuta</i> spp. [Except for native species]
Hydrilla	<i>Hydrilla verticillata</i>
Broomrape	<i>Orobanche</i> spp. [Except for native species]
Wavyleaf basketgrass	<i>Opismenus hirtellus</i>

Class B Noxious Weeds

Bull thistle	<i>Cirsium vulgare</i>
Canada thistle	<i>Cirsium arvense</i>
Musk thistle	<i>Carduus nutans</i>
Johnson grass	<i>Sorghum halepense</i>
Mile-a-minute	<i>Persicaria perfoliata</i>
Multiflora rose	<i>Rosa multiflora</i>
Purple Loosestrife	<i>Lythrum salicaria</i>
Shattercane	<i>Sorghum bicolor</i>
Poison hemlock	<i>Conium maculatum</i>

Class C Noxious Weeds

Refer to USDA Federal Noxious Weed List (attached); Class C includes any species from this list not categorized as Class A or B

Appendix B

Noxious Weed Identification Sheets

Invasive Plants in Pennsylvania

Canada Thistle

Cirsium arvense



Steve Dewey, Utah State University
www.forestryimages.org

Description:

Canada thistle is an erect herbaceous perennial with an extensive creeping rootstock. Its leaves are irregularly lobed with spiny, toothed margins. Rose-purple or sometimes white flower heads appear in terminal clusters from June through October. The small seeds have feathery plumes.

Biology and Spread:

Canada thistle produces an abundance of feathery seeds, which are quickly dispersed in the wind. The seeds can remain viable in the soil for up to 20 years or more. The fibrous taproot is capable of sending out lateral roots, which sprout shoots at frequent intervals.

Background:

Canada thistle was probably introduced into the United States by accident in the early 1600s. By 1954, it had been declared a noxious weed in 43 states. It is considered one of the most tenacious and economically important agricultural weeds and is becoming increasingly recognized as a problem in natural areas.



Steve Dewey, Utah State University
www.forestryimages.org

Ecological Threat:

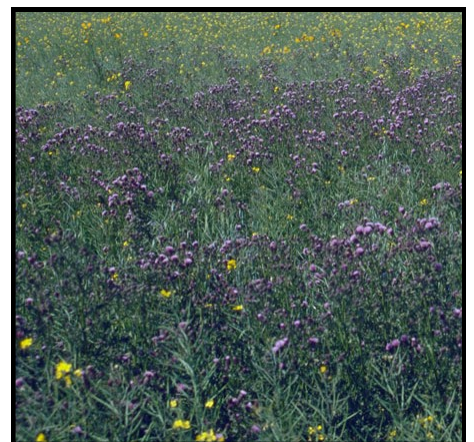
Once established in an area, Canada thistle crowds out and replaces native plants, changing the structure and species composition of plant communities and reducing diversity. This thistle outcompetes native plants through shading, competition for soil resources and possibly through the release of toxic allelochemicals.

Range:

Despite its name, Canada thistle is native to temperate regions of Eurasia. In North America, it is distributed throughout Canada and the northern United States, from northern California to Maine and south to Virginia.

Habitat:

This plant does best in open and disturbed upland areas, but also invades wet places with fluctuating water levels, such as stream bank meadows. It is commonly found in barrens, glades, meadows, prairies, fields, pastures and waste places.



Alec McClay, McClay Ecoscience
www.forestryimages.org

How to Control this Species:

Because Canada thistle is a perennial and spreads primarily by its root system, the entire plant must be destroyed for effective control.

Control efforts may be more successful when Canada thistle is under environmental stress, such as during droughts and floods, or after a very severe winter.

Canada thistle is stubborn and difficult to remove. Management practices that limit soil disturbance and encourage diverse native plant communities will help prevent establishment of this species.

Physical

For light infestations, black plastic sheeting can be used to smother this thistle. Repeated and frequent pulling or hand-cutting will eventually starve underground stems. This should be performed at least three times each season.

Mowing does not kill Canada thistle unless repeated monthly for up to four years. This method is not recommended for natural areas.

Late spring burns, between May and June, are detrimental to this invasive.

Chemical

In areas interspersed with desirable native plants, targeted application of a systemic herbicide, such as glyphosate, works well. For extensive infestations in disturbed areas, a broad application may be more effective. Repeated applications are usually necessary in order to exhaust the seed bank.

Herbicide treatment is best done in late summer or fall when plants are in the rosette stage.

Varying the type of herbicide used will prevent clone colonies from becoming resistant.

Look-A-Likes:

Native species of thistle (*Cirsium* sp.), some of which are rare, could be confused with Canada thistle. Before control is attempted, the thistle species in question should be accurately identified.



Field Thistle

Chris Evans, River to River CWMA
www.forestryimages.org

References:

Center for Invasive Species and Ecosystem Health:
<http://www.invasive.org/browse/subinfo.cfm?sub=2792>

Plant Conservation Alliance's Alien Plant Working Group:
<http://www.nps.gov/plants/alien/fact/ciar1.htm>

For More Information:

DCNR Invasive Species Site: <http://www.dcnr.state.pa.us/conservationscience/invasivespecies/index.htm>

DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers:
http://www.dcnr.state.pa.us/forestry/invasivetutorial/canada_thistle.htm

Invasive Plants in Pennsylvania

Bull Thistle

Cirsium vulgare (Savi) Ten.



Photo: Western New Mexico University, Dept. of Nat. Sci. & Dale A. Zimmerman Herbarium

Description:

Stems grow up to seven feet tall and have lance shaped, hairy leaves that are three to 12 inches long. There are lobes on each leaf, and they are tipped with stout spines. Flower heads are purple and one to two inches across. They appear from June to September.

Biology and Spread:

Bull thistle reproduces and spreads exclusively by seed. Plants produce 100 to 300 seeds per flower head, with up to 400 flower heads per plant. Seeds exhibit features that are suited for wind dispersal. Roughly 90 percent of seeds land within a distance of 1.5 times the height of the parent plant, however up to 10 percent of seeds produced do travel over 80 feet.

Background:

Thought to have been introduced to eastern North America during colonial times and western North America during late 1800s.

Range:

This plant is native to Europe, western Asia and northern Africa. It can now be found on every continent except Antarctica, and it is present in all 50 states in the U.S., although it is not as common in the southern states as it is in the north.



Photo: Steve Dewey, Utah State U., www.invasive.org

Ecological Threat:

Bull thistle has the potential to form dense thickets that displace and prevent native vegetation from establishing. The spiny features of the plant also make it unpalatable to wildlife and livestock, reducing forage land available.

Habitat:

This plant has the ability to become established in a wide variety of areas, especially ones that have been disturbed, like clearcuts, riparian areas and pastures.



Photo: Steve Dewey, Utah State U., www.invasive.org

How to Control this Species:

Prevention

Prevention should be the first goal in any control efforts for this plant. Equipment and gear need to be extensively cleaned after use in an infested area, to prevent seed transport. Use of weed-free seed should also be conducted.

Manual and Mechanical

Manual removal of plants via hand pulling, tilling and hoeing can be effective, but care should be taken to conduct these measures before flowering, in order to prevent seed production and spread. Slicing off the root crown has proven to be very effective at controlling this plant.

Chemical

Herbicides are most effective at controlling infestations when incorporated as part of a long-term, multifaceted control effort. Herbicide applications should be optimally timed in order to have the greatest effect.

Regardless of the control measures taken, reestablishing competitive, desirable vegetation to the site is critical.

Look-A-Likes:

Native species of thistle (*Cirsium* sp.), some of which are rare, could be confused with bull thistle. Before control is attempted, the thistle species in question should be accurately identified.

References:

Zouhar, Kris. 2002. *Cirsium vulgare*. In: *Fire Effects Information System*. U.S. Department of Agriculture, Forest Service, Rocky Mountain Research Station, Fire Sciences Laboratory:

<http://www.fs.fed.us/database/feis>

Center of Invasive Species and Ecosystem Health: www.invasive.org

For More Information:

DCNR Invasive Species Site: <http://www.dcnr.state.pa.us/conservationscience/invasivespecies/index.htm>

DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers: http://www.dcnr.state.pa.us/forestry/invasivetutorial/canada_thistle.htm

Plant Invaders of Mid-Atlantic Natural Areas, National Park Service, <http://www.nps.gov/plants/alien/pubs/midatlantic/midatlantic.pdf>

Invasive Plants Field and Reference Guide, U.S. Forest Service: http://na.fs.fed.us/pubs/misc/ip/ip_field_guide.pdf



Chris Evans, River to River CWMA
www.forestryimages.org



Invasive Plants in Pennsylvania

Poison Hemlock

Conium maculatum



Pedro Tenorio-Lezama
www.forestryimages.org

Background:

In the 1800s, poison hemlock was brought to the United States from Europe as an ornamental. In ancient times, it was probably used to poison Socrates, a famous Greek philosopher.

Range:

Poison hemlock is native to Europe, western Asia and North Africa. It is now widespread throughout much of North America. It has also been introduced to other continents, such as South America and Australia.

Description:

Poison hemlock is a biennial herb with hollow, purple-spotted stems that can reach eight feet in height. Its finely dissected leaves emit a foul, parsnip-like odor when crushed. Plants begin as a rosette of leaves and flower in the second year of growth. The small, white flowers are borne in umbrella-shaped clusters.



Eric Coombs, Oregon Dept. of Ag.
www.forestryimages.org

Habitat:

This plant commonly occurs in dense stands along roadsides, field margins, irrigation ditches and waste areas. It also invades native plant communities in riparian woodlands, open floodplains and along stream banks.

Biology and Spread:

A single poison hemlock plant can produce over 30,000 seeds. These seeds can adhere to farm machinery, vehicles, fur and clothing, as well as be carried by water, and to a limited extent, wind. Poison hemlock is capable of rapid establishment, particularly in disturbed sites.

Ecological Threat:

Poison hemlock can be a tenacious weed, particularly in moist sites. As a pioneer species, it quickly colonizes disturbed sites, displacing natives. All parts of the plant, especially the seeds, are extremely poisonous to humans and livestock.



John D. Byrd, Mississippi State U.
www.forestryimages.org

How to Control this Species:

Physical

Hand-pulling works best for wet soils with small infestations. Because poison hemlock is not a perennial, removal of the entire root system is not necessary.

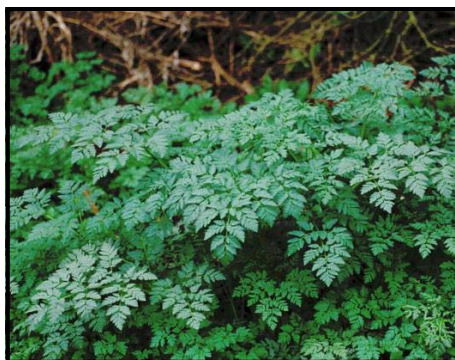
Mowing or cutting the plant close to the ground just before flowering is often effective, but may require retreatment if new growth is produced at the base.

Poison hemlock remains toxic for several years after being pulled. Ensure that the material is kept out of reach of children and wildlife.

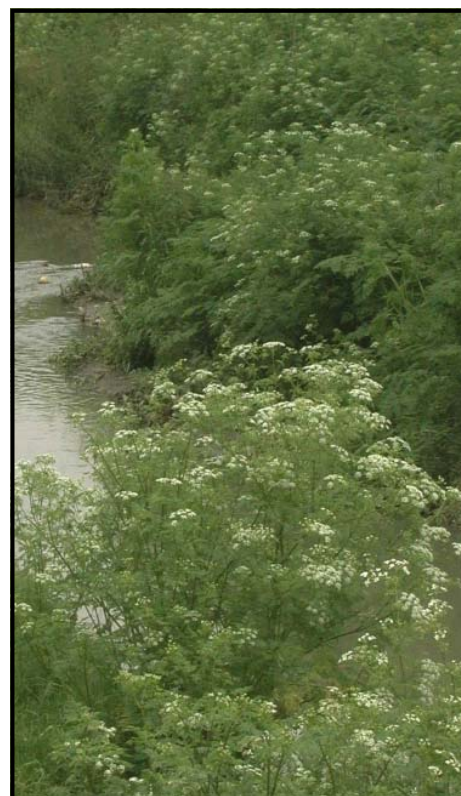
Chemical

The application of herbicides, such as glyphosate and 2,4-D can effectively control large infestations.

Complete eradication may be difficult if a viable seedbank is present.



Barry Rice, sarracenia.com
www.forestryimages.org



Pedro Tenorio-Lezama
www.forestryimages.org

Look-A-Likes:

Poison hemlock is sometimes confused with the invasive giant hogweed (*Heracleum mantegazzianum*) and our native water hemlock (*Cicuta maculata*). Deaths have occurred from mistaking the roots for wild carrots.



Steve Dewey, Utah State University
www.forestryimages.org

References:

Center for Invasive Species and Ecosystem Health:

<http://www.invasive.org/browse/subinfo.cfm?sub=4365#maps>

USDA Forest Service: <http://www.invasive.org/weedcd/pdfs/wow/poison-hemlock.pdf>

For More Information:

DCNR Invasive Species Site: <http://www.dcnr.state.pa.us/conservationscience/invasivespecies/index.htm>

DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers:
http://www.dcnr.state.pa.us/forestry/invasivetutorial/poison_hemlock.htm

Invasive Plants in Pennsylvania

Purple Loosestrife

Lythrum salicaria



Richard Old, XID Services, Inc.
www.forestryimages.org

Description:

Purple loosestrife is a perennial herb with square, woody stems, which may grow anywhere from four to 10 feet high, depending on conditions. Its lance-shaped leaves occur in opposite or whorled arrangements. Magenta-colored flower spikes are present throughout much of the summer.



Eric Coombs, Oregon Dept. of Ag.
www.forestryimages.org

Biology and Spread:

With an extended flowering season, from June to September, and an unusually high number of flowering stems, each purple loosestrife plant is capable of producing two to three million seeds per year. Research has shown that cultivars, advertised as sterile, are capable of producing viable seed. This plant can also reproduce vegetatively by underground stems at a rate of one foot per year.

Ecological Threat:

An enthusiastic invader of wetlands, purple loosestrife outcompetes native plants, including some federally endangered orchids, forming dense homogeneous stands. These monocultures reduce habitat for waterfowl, clog waterways, disrupt nutrient cycling and collect debris, eventually displacing the entire wetland.



Agriculture and Agri-Food Canada
www.forestryimages.org

Background:

Purple loosestrife was intentionally introduced into North America in the early 1800s as an ornamental, as well as accidentally by way of discarded ship ballast. It is now banned as a noxious weed in most states.

Range:

Native to Eurasia, purple loosestrife can now be found throughout much of the United States, especially in the northern and western regions.

Habitat:

Purple loosestrife prefers open wetlands, and is capable of invading freshwater wet meadows, tidal and non-tidal marshes, river and stream banks, pond edges, reservoirs and ditches. It is a hardy plant that can tolerate a range of soil pH, as well as drought.

How to Control this Species:

Prevention

Early detection and prevention are the best approaches to managing purple loosestrife. Monitoring watersheds yearly to identify new infestations is critical, and can be most easily conducted in late July and August when the plant is in full bloom.

Clean seed and plant parts from animals, equipment and clothing before entering wetland areas.

Prevent nearby infestations from going to seed. Use native competitors as barriers.

Physical

Hand-pulling is only effective for seedlings with small roots.

Mowing is not recommended, but may reduce the production of seeds.

Flooding kills seedlings; established plants must be inundated for weeks. Unfortunately, this also kills desirable vegetation.

The site may need to be replanted with native, competitive vegetation.

Chemical

Glyphosate is effective against purple loosestrife. Be sure to use an herbicide permitted for wetland use. Herbicides can be applied directly to cut stems to reduce collateral damage.

Biocontrol

Although they will not eradicate purple loosestrife, biocontrols can reduce the severity of an infestation. Four species of beetles from Europe, which are fairly host-specific on purple loosestrife, are currently available for control efforts.

Look-A-Likes:

From a distance, purple loosestrife may resemble some native flowering plants, such as blazing star (*Liatris* sp.) and obedient plant (*Physostegia virginiana*). These also make great native garden alternatives.



Blazing Star

John D. Byrd, Mississippi State University
www.forestryimages.org

References:

Center for Invasive Species and Ecosystem Health:

<http://www.invasive.org/browse/subinfo.cfm?sub=3047>

Plant Conservation Alliance's Alien Plant Working Group:

<http://www.nps.gov/plants/alien/fact/lysa1.htm>

University of Nevada Cooperative Extension:

<http://www.unce.unr.edu/publications/files/nr/2002/>

For More Information:

DCNR Invasive Species Site: <http://www.dcnr.state.pa.us/conservationscience/invasivespecies/index.htm>

DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers:

http://www.dcnr.state.pa.us/forestry/invasivetutorial/Purple_loosestrife.htm

Invasive Plants in Pennsylvania

Mile-a-Minute

Persicaria perfoliata



Photo: Leslie Mehrhoff, U. of Connecticut,
www.invasive.org

Background:

Also known as devil's tear-thumb, mile-a-minute has been introduced into the U.S. from the Philippines several times between the late 1800s and the 1930s. It arrived in Pennsylvania in contaminated nursery stock in York.

Range:

A native of eastern Asia, this vine is not yet widespread in the U.S. but is very common in the southern two-thirds of Pennsylvania, as well as parts of WV, VA, MD, DE, NJ, NY, CT, MA, RI and NH.

Description:

This is an herbaceous, annual vine with delicate, highly branched stems that are covered by small, curved spines. The alternate leaves are triangular, light green, one to three inches wide and barbed on the underside. Round leaf-like structures called ocreae surround the stem. It is from there that the inconspicuous flowers and fruits arise. From mid-July through the first frost, green fruits appear, turning a metallic blue color as the season goes on.



Photo: Jessica Sprajcar, DCNR

Habitat:

This plant readily colonizes disturbed areas along forest edges, wetlands, stream banks and roadsides. It needs regular sunlight to thrive and prefers high soil moisture.

Biology and Spread:

Its fast growth is one way that the plant spreads, but its seeds are the primary means. Birds and other wildlife eat the fruits and spread the seeds in their droppings. Seeds are also buoyant for up to nine days in water and can be spread by streams and floods.

Ecological Threat:

Because this plant can grow up to six inches a day, it can quickly smother native vegetation and climb into the tree canopy where it restricts light availability to plants below. It can be a pest plant on tree farms and for horticultural crops where the soil is not regularly tilled.



Photo: Leslie Mehrhoff, U. of Connecticut,
www.invasive.org

How to Control this Species:

Manual and Mechanical

Hand-pulling of vines is possible, especially when the soil is wet, but be sure to wear thick gloves. Removal should be done prior to fruit formation. Repeated mowing will prevent the plant from flowering and thus reduce or eliminate fruit and seed production. Monitor the site for several years to ensure no seeds germinate.

Chemical

A systemic herbicide like glyphosate will work on mile-a-minute, especially when used with a surfactant that will help to penetrate the leaves' waxy coating. Apply the herbicide in the summer, before fruits appear.



Rhinocominus latipes

Photo: NJ Dept. of Agriculture

Biocontrol

A weevil, *Rhinocominus latipes*, is being used on various test plots in Pennsylvania and elsewhere to control mile-a-minute. These small insects feed on the leaves and bore into the stems. While they will not completely eliminate the plant they help keep it in check and reduce fruit production.

Look-a-Likes:

There are several other vines with triangular-shaped leaves that may be confused with mile-a-minute, including halbard-leaved tearthumb (*Polygonum arifolium*), climbing false buckwheat (*Polygonum scandens*), wild morning glory (*Ipomoeae pupurea*) and hedge bindweed (*Calystegia sepium*). The presence of spines and ocreae will let you know that it is indeed mile-a-minute.



Photo: Theodore Webster, USDA,
www.forestryimages.org

References:

Center for Invasive Species and Ecosystem Health:

<http://www.invasive.org/browse/subinfo.cfm?sub=3065>

Invasive Exotic Plant Tutorial for Natural Lands Managers:

http://www.dcnr.state.pa.us/forestry/invasivetutorial/mile_a_minute.htm

University of Delaware, College of Agriculture & Natural Resources, Biological Control of Mile-a-Minute Weed: <http://ag.udel.edu/enwc/research/biocontrol/mileaminute.htm>

Massachusetts Introduced Pests Outreach Project:

<http://massnrc.org/pests/mamreport.aspx>

For More Information:

Plant Invaders of Mid-Atlantic Natural Areas, National Park Service:

<http://www.nps.gov/plants/alien/pubs/midatlantic/midatlantic.pdf>

Invasive Plants Field and Reference Guide, U.S. Forest Service:

http://na.fs.fed.us/pubs/misc/ip/ip_field_guide.pdf

Invasive Plants in Pennsylvania

Multiflora Rose

Rosa multiflora



Leslie J. Mehrhoff, U. of Connecticut
www.forestryimages.org

Background:

Multiflora rose was introduced into the United States as ornamental rootstock from Japan in 1866. Beginning in the 1930s, the U.S. Soil Conservation Service promoted it for use in erosion control and livestock fencing. It was also encouraged in wildlife plantings and as a crash barrier along highways. Recognition of its tenacious and unstoppable growth habit came too late, and it is now considered a noxious weed in many states.

Range:

Native to Asia, multiflora rose now occurs throughout most of the United States, especially the eastern half.

Description:

Multiflora rose is a dense, thorny shrub, reaching up to 15 feet in height, with arching canes (stems) that are capable of rambling up trees. Its leaves are pinnately compound, divided into seven to nine leaflets, and finely serrate. Clusters of fragrant white to pink flowers appear in May or June. Small bright red hips (fruit) develop during the summer and remain on the plant through winter.



James H. Miller, USDA Forest Service
www.forestryimages.org

Habitat:

This invasive shrub has a wide tolerance for various soil, moisture and light conditions. It can be found in dense woods, along stream banks and roadsides, and in open fields and prairies.

Biology and Spread:

It is estimated that a single plant may produce a million seeds per year, which may remain viable in the soil for up to 20 years. The hips are readily eaten by birds, which are the primary seed dispersers. New plants can also be formed by rooting from the tips of canes touching the ground.

Ecological Threat:

Multiflora rose forms impenetrable thickets that exclude native plant species. This shrub grows very prolifically in riparian areas, where its inedible leaf litter can change the composition of the aquatic macroinvertebrate community. Its occasional habit of climbing can weigh down trees, making them susceptible to breakage.



James H. Miller, USDA Forest Service
www.forestryimages.org

How to Control this Species:

Physical

Frequent cutting or mowing, three to six times per growing season, for two to four years, is effective in achieving high mortality. Be careful—the strong thorns have been known to puncture rubber tires.

Scattered populations may be eliminated by complete removal of the plants. Be sure to remove all root material because this shrub readily re-sprouts.

In areas where multiflora rose is detected early, prescribed fire may limit its establishment.

Chemical

Application of herbicides, such as glyphosate or triclopyr, on freshly cut stems is an effective control method since it destroys the root system and prevents re-sprouting. This may be done during the dormant period, which reduces the likelihood of damaging desirable species.

A foliar spray of fosamine can be used from July through September, but die-back will not be apparent until the following summer. Fosamine will only affect woody species.

Biological

Biological control is currently under investigation. Rose-rosette disease, a native viral pathogen, is spread by a mite, and is slowly spreading eastward from the west. The European rose chalcid, a seed-infesting wasp, promises to reduce seed viability. Unfortunately, both of these measures have the potential to impact native rose species.



Leslie J. Mehrhoff, U. of Connecticut
www.forestryimages.org

Look-A-Likes:

Multiflora rose could easily be confused with other rose species (both native and non-native), especially when not in bloom. This is a concern, since some native species are of conservation interest.



Catherine Herms, Ohio State University
www.forestryimages.org

References:

Plant Conservation Alliance's Alien Plant Working Group:
<http://www.nps.gov/plants/alien/fact/romu1.htm>

Wisconsin Department of Natural Resources:
<http://dnr.wi.gov/invasives/fact/rose.htm>

For More Information:

DCNR Invasive Species Site: <http://www.dcnr.state.pa.us/conservationscience/invasivespecies/index.htm>

DCNR Invasive Exotic Plant Tutorial for Natural Lands Managers:
http://www.dcnr.state.pa.us/forestry/invasivetutorial/Multiflora_rose.htm

Appendix C
Invasive Insect Species Identification Sheets

Emerald Ash Borer



A beetle from Asia, *Agrilus planipennis* Fairmaire (Coleoptera: Buprestidae), was identified in July 2002 as the cause of widespread ash (*Fraxinus* spp.) tree decline and mortality in southeastern Michigan and Windsor, Ontario, Canada. Larval feeding in the tissue between the bark and sapwood disrupts transport of nutrients and water in a tree, eventually causing branches and the entire tree to die. Tens of millions of ash trees in forest, rural, and urban areas have already been killed or are heavily infested by this pest.

A. planipennis has been found throughout Michigan, across much of Ohio, and in parts of Indiana, Illinois, Maryland, Missouri, Pennsylvania, Virginia, West Virginia and Wisconsin. Infestations have also been found in more areas of Ontario and in the province of Quebec. The insect is likely to be found in additional areas as detection surveys continue. Evidence suggests that *A. planipennis* is generally established in an area for several years before it is detected.

The broad distribution of this pest in the United States and Canada is primarily due to people inadvertently transporting infested ash nursery stock, unprocessed logs, firewood, and other ash commodities. Federal and state quarantines in infested states now regulate transport of these products.

Identification

Adult beetles are generally larger and brighter green (Fig. 1) than the native North American *Agrilus* species. Adults are slender, elongate, and 7.5 to 13.5 mm long. Males are smaller than females and have fine hairs, which the females lack, on the ventral side of the thorax. Adults are usually bronze, golden, or reddish green overall, with darker, metallic emerald green wing covers. The dorsal side of the abdomen is metallic purplish red and can be seen when the wings are spread (Fig. 2). The prothorax, the segment behind the head and to which the first pair of legs is attached, is slightly wider than the head and the same width as the base of the wing covers.

Larvae reach a length of 26 to 32 mm, are white to cream-colored, and dorso-ventrally flattened (Fig. 3). The brown head is mostly retracted into the prothorax, and only the mouthparts are visible. The abdomen has 10 segments, and the last segment has a pair of brown, pincer-like appendages.

Biology

A. planipennis generally has a 1-year life cycle. In the upper Midwest, adult beetles begin emerging in May or early June. Beetle activity peaks between mid June and early July, and continues into August. Beetles probably live for about 3 weeks, although some have survived for more than 6 weeks in the laboratory. Beetles generally are most active during the day, particularly when it is warm and sunny. Most beetles appear to remain in protected locations in bark crevices or on foliage during rain or high winds.

Throughout their lives beetles feed on ash foliage, usually leaving small, irregularly shaped patches along the leaf margins. At least a few days of feeding are needed before beetles mate, and an additional 1 to 2 weeks of feeding may be needed before females begin laying eggs. Females can mate multiple times. Each female probably lays 30-60 eggs during an average lifespan, but a long-lived female may lay more than 200 eggs. Eggs are deposited individually in bark crevices or under bark flaps on the trunk or branches, and soon darken to a reddish brown. Eggs hatch in 7 to 10 days.

After hatching, first instar larvae chew through the bark and into the phloem and cambial region. Larvae feed on phloem for several weeks, creating serpentine (S-shaped) galleries packed with fine sawdust-like frass. As a larva grows, its gallery becomes progressively wider (Fig. 4). Beetle galleries often etch the outer sapwood. The length of the gallery generally ranges from 10 to 50 cm. Feeding is usually completed in autumn.

Prepupal larvae overwinter in shallow chambers, roughly 1 cm deep, excavated in the outer sapwood or in the bark on thick-barked trees. Pupation begins in



Figure 1. Adult emerald ash borer.



Figure 2. Purplish red abdomen on adult beetle.



Figure 3. Second, third, and fourth stage larvae.



Figure 4. Gallery of an emerald ash borer larva.



Figure 5. D-shaped hole where an adult beetle emerged.



Figure 6. Jagged holes left by woodpeckers feeding on larvae.



Figure 7. Ash tree killed by emerald ash borer. Note the serpentine galleries.



Figure 8. Epicormic branching on a heavily infested ash tree.

late April or May. Newly eclosed adults often remain in the pupal chamber or bark for 1 to 2 weeks before emerging head-first through a D-shaped exit hole that is 3 to 4 mm in diameter (Fig. 5).

Studies in Michigan indicate 2 years may be required for *A. planipennis* to develop in newly infested ash trees that are relatively healthy. In these trees, many *A. planipennis* overwinter as early instars, feed a second summer, overwinter as prepupae, and emerge the following summer. In trees stressed by physical injury, high *A. planipennis* densities, or other problems, all or nearly all larvae develop in a single year. Whether a 2-year life cycle will occur in warmer southern states is not yet known.

Distribution and Hosts

A. planipennis is native to Asia and is found in China and Korea. It is also reported in Japan, Mongolia, the Russian Far East, and Taiwan. In China, high populations of *A. planipennis* occur primarily in *Fraxinus chinensis* and *F. rhynchophylla*, usually when those trees are stressed by drought or injury. Other Asian hosts (*F. mandshurica* var. *japonica*, *Ulmus davidiana* var. *japonica*, *Juglans mandshurica* var. *sieboldiana*, and *Pterocarya rhoifolia*) may be colonized by this or a related species.

In North America *A. planipennis* has attacked only ash trees. Host preference of *A. planipennis* or resistance among North American ash species may vary. Green ash (*F. pennsylvanica*) and black ash (*F. nigra*), for example, appear to be highly preferred, while white ash (*F. americana*) and blue ash (*F. quadrangulata*) are less preferred. At this time all species and varieties of native ash in North America appear to be at risk from this pest.

Signs and Symptoms

It is difficult to detect *A. planipennis* in newly infested trees because they exhibit few, if any, external symptoms. Jagged holes excavated by woodpeckers feeding on late instar or prepupal larvae may be the first sign that a tree is infested (Fig. 6). D-shaped exit holes left by emerging adult beetles may be seen on branches or the trunk, especially on trees with smooth bark (Fig. 5). Bark may split vertically over larval feeding galleries. When the bark is removed from infested trees, the distinct, frass-filled larval galleries that etch the outer sapwood and phloem are readily visible (Fig. 4 and Fig. 7). An elliptical area of discolored sapwood, usually a result of secondary infection by fungal pathogens, sometimes surrounds galleries.

As *A. planipennis* densities build, foliage wilts, branches die, and the tree canopy becomes increasingly thin. Many trees appear to lose about 30 to 50 percent of the canopy after only a few years of infestation. Trees may die after 3 to 4 years of heavy infestation (Fig. 7). Epicormic shoots may arise on the trunk or branches of the tree (Fig. 8), often at the margin of live and dead tissue. Dense root sprouting sometimes occurs after trees die.

A. planipennis larvae have developed in branches and trunks ranging from 2.5 cm (1 inch) to 140 cm (55 inches) in diameter. Although stressed trees are initially more attractive to *A. planipennis* than healthy trees are, in many areas all or nearly all ash trees greater than 3 cm in diameter have been attacked.

Resources

For more information on the emerald ash borer and related topics...

- Visit the following Web sites:

Multi-agency Emerald Ash Borer Web Site:

www.emeraldashborer.info

USDA Forest Service: www.na.fs.fed.us/fhp/eab/

USDA Animal and Plant Health Inspection Service:

www.aphis.usda.gov/plant_health/

- Contact your state Department of Agriculture, State Forester, or Cooperative Extension Office.

Prepared by:

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Photo credits:

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Edward Czerwinski, Ontario Ministry of Natural Resources, www.forestryimages.org



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Forest Health Fact Sheet

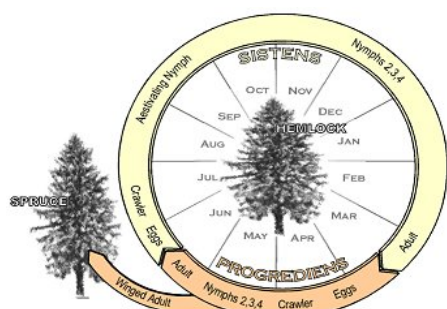
Hemlock Woolly Adelgid

Adelges tsugae, the hemlock woolly adelgid, is a fluid-feeding insect that feeds on hemlock trees throughout eastern North America, including Pennsylvania. The egg sacs of these insects look like the tips of cotton swabs clinging to the undersides of hemlock branches.

Hemlock woolly adelgid was introduced from Asia into the Pacific Northwest in 1924. It was probably introduced into the northeastern US in the 1950's, and it was first discovered in Pennsylvania in 1967. This insect has been damaging hemlock ever since, and it is spreading. To date, 49 counties in the eastern two-thirds of PA have been infested with this insect.



Look for hemlock woolly adelgid on the undersides of lower branches

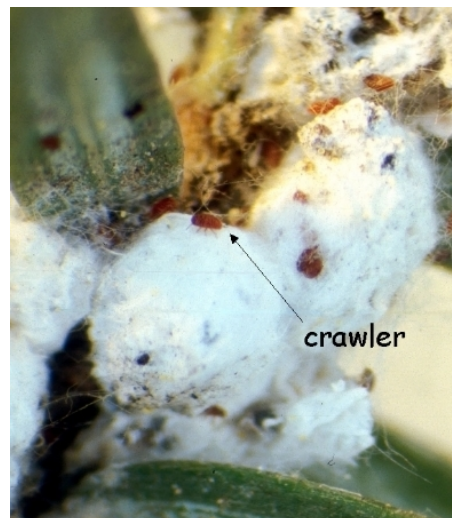


Life Cycle, Courtesy of Vince D'Amico and Mike Montgomery

Hemlock woolly adelgid has two generations per year in Pennsylvania. All populations are made up of females that reproduce asexually. In early spring, overwintering females lay between 100 and 300 eggs in the woolly egg sacs beneath the branches.

Mobile larvae, known as crawlers, emerge from the eggs in April or May to search for suitable feeding sites. Wind, birds and mammals often spread crawlers to nearby hemlocks. Once settled at the base of hemlock needles, crawlers become immobile nymphs which

feed and mature into wingless or winged adult females by early summer. The winged form will die after searching for a suitable spruce tree that is not found in North America. The wingless form lays another 100 to 300 eggs on hemlock. Crawlers emerge from these eggs to search for suitable feeding sites. Once settled, the hemlock woolly adelgid becomes dormant until October or November, when it resumes development. Feeding continues throughout the winter and early spring.



Adelgid larvae emerge from eggs

Eastern hemlock (Pennsylvania's state tree) and Carolina hemlocks (found further south in the Smokey Mountain sections of the Appalachians) are more susceptible to hemlock woolly adelgid damage than Asian and western hemlock trees due to feeding tolerance and predators that protect the latter species. Hemlock woolly adelgid sucks fluid from the base of hemlock needles. It may also inject toxins into the tree as it feeds, accelerating needle drop and branch dieback. Although some trees die within four years, trees often persist in a weakened state for many years. Hemlocks that have been affected by hemlock woolly adelgid often have a grayish-green appearance (hemlocks naturally have a shiny, dark green color).

Other factors can influence the impact of the hemlock woolly adelgid. Other insects, such as elongate hemlock scale, hemlock borer, and spittlebugs, which are also found on hemlock, can compound the impact of hemlock woolly adelgid. Drought and fungi, such as *Fabrella* or *Korfia tsugae* can weaken hemlock and cause it to become more susceptible to insect damage. Low winter temperatures, cold snaps (episodes of freezing and thawing), and heavy thunderstorms can reduce populations of the hemlock woolly adelgid. Particularly in the mountains, it is not uncommon to find hemlocks where the insect has been killed on the top third of the trees, where it's colder and windier, but survive on the bottom two-thirds. On the other hand, mild winters can result in sharp increases in hemlock woolly adelgid populations.

PA DCNR uses integrated pest management (IPM) principles to manage hemlock woolly adelgid. IPM relies on survey and monitoring of the insect and its hemlock host. It involves using a variety of management techniques,

such as biological, chemical, cultural, and silvicultural control to reduce the populations to less damaging levels. The choice of control method will vary depending on the site and other circumstances of each situation. Unfortunately, eradication is not the objective, because hemlock woolly adelgid is already firmly established in our state.

Survey and Monitoring PA DCNR is currently attempting to map ecologically significant hemlock stands in our state, in order to detect new infestations, focus our control efforts and predict areas most vulnerable to hemlock woolly adelgid. We are using a variety of remote-sensing and ground-based techniques to accomplish this objective. Remote sensing technologies are also being evaluated for monitoring hemlock woolly adelgid. DCNR is cooperating with Rutgers to expand an algorithm developed for using Landsat imagery to detect changes in hemlock health in NJ to PA. DCNR and USFS are working with cooperators to determine if hyperspectral images taken from helicopters can detect new infestations along the leading edge and in isolated patches.

Biological control is the release of natural enemies that attack the pest. Biological controls include predators, disease organisms (fungi, bacteria, and viruses), and parasitoids (predators eat their prey, and parasitoids reproduce in their victims). Unfortunately, there are no known parasitoids of the family Adelgidae, to which hemlock woolly adelgid belongs. Pennsylvania is a key participant in national efforts to use biological control towards the management of the hemlock woolly adelgid. Unlike chemical control, biological control is a long-term, permanent strategy to managing hemlock woolly adelgid throughout Pennsylvania. In spite of these benefits, biocontrol is NOT usually recommended for landowner use. In the case of beetles, the high cost (around \$3 each), large numbers of beetles required for control (thousands per site), the tendency of biocontrols to fly away from the initial release site, and the lengthy time (5-10 years) expected for noticeable results can make this method impractical. It is our hope that the biological control agents we release will, in time, protect hemlocks in ornamental situations as well as forests.

Beetles to the Rescue

Biocontrol of hemlock woolly adelgid is used in forest situations, on vigorous trees with accessible lower branches that are infested with hemlock woolly adelgid. It is likely that PA DCNR will eventually establish several natural enemies that will work together to reduce the impact of hemlock woolly adelgid.



Pseudoscymnus tsugae (PT) is a pinhead-sized, specialized, black lady beetle discovered feeding on hemlock woolly adelgid in Japan in 1992. Having completed several years of successful tests in cooperation with the U. S. Forest Service, DCNR's Bureau of Forestry has embarked on a statewide PT release effort. PT only eats hemlock woolly adelgid, balsam woolly adelgid, and pine bark adelgid. Each larva eats about 500 eggs or 50 -100 immature adelgids, known as nymphs. PT females lay up to 300 eggs in March and April, during peak egg-laying of hemlock woolly adelgid. The beetles have a second generation in June around the same time as the second adelgid generation. Adult PT feed on

dormant young adelgids during the summer. Releases of PT are focused on hemlocks along the leading edge of HWA infestation. 176,387 beetles have been released at 50 sites in 23 counties. DCNR staff uses a plastic bat to beat branches over a white sheet to see if PT has established at sites in the years following release. This lady beetle should not be viewed as a cure-all, since its effectiveness is still in the testing stages. However, the data show some encouraging signs so far. To date, we have recovered 642 beetles. Adult recovery indicates overwintering success, and larval recovery indicates successful field reproduction.



Pseudoscymnus on a researcher's finger.

The lady beetles are literally grown in lady beetle farms in Pennsylvania and New Jersey, and are used for testing in hemlock sites all over the eastern seaboard. In order to make the biocontrol program more affordable, Forest Pest Management has set up a cooperative agreement with New Jersey Department of Agriculture that involves sending tons of infested hemlock foliage a year to be used for rearing hemlock woolly adelgid. In return, NJDA provides us with a substantial percentage of their PT beetles each year.

An ironic problem with the lady beetles, unfortunately, is their voracious appetite for the hemlock woolly adelgid eggs. If they are not fed enough, they tend to not multiply as quickly and can fly off in search of better feeding

grounds. However, the lady beetles have proven to be quite effective in the test sites at devouring the woolly adelgid, perhaps improving the outlook for threatened hemlocks and reducing long-term pesticide use.



More beetles

Laricobius nigrinus (LN): is a beetle that is native to the western North America where it preys on hemlock woolly adelgid on western hemlock. *Laricobius* beetles only feed on woolly adelgids. LN adults lay eggs in early spring on overwintering HWA nymphs. Larvae emerge and feed on HWA until they mature in spring, when they enter the soil to pupate. Adults remain dormant in the soil until fall. LN beetles prefer HWA to other adelgids, and it can only complete its development on this species. PA released 300 LN beetles at a site in Huntingdon County in December 2003, as part of a cooperative study with Virginia Polytechnic Institute. More releases are planned in the eastern US over the next three years. Other lady beetles are also being tested for effectiveness against

hemlock woolly adelgid in Connecticut and New Jersey. A leading candidate is the *Scymnus sinuanodulus* (SS) lady beetle from China. SS is yet another predator that feeds on woolly adelgid eggs. In addition to these, Connecticut is currently working with yet another type of Chinese lady beetle, *S. ningshangiensis*. Once laboratory testing is complete, DCNR's Bureau of Forestry may add one or more of these new beetles to the hemlock woolly adelgid arsenal.

No, not that Lady beetle



Some folks who hear about DCNR's lady beetle releases are actually upset! They have had problems with another lady beetle, the **Asian Multicolored Lady Beetle**, which congregates on and in peoples' homes on sunny spring and fall days. Having the scientific name of *Harmonia axyridis* (HA), this Asian lady beetle is *huge* compared to the PT or *Scymnus* lady beetles-as big around as a pencil eraser-and is very colorful. Over one hundred PT beetles could hitch a ride on the back of one HA lady beetle! HA may be orange, brown, red, or yellow and could have black spots. It first was released by the U.S. Department of Agriculture in the 1920s to control aphids (another sucking insect) on agricultural crops. Once established, this beetle spread rapidly, until by the early '90s it could be found nearly everywhere. Attracted to warm, sheltered spots in the fall, HA prefers light-colored, sunlit houses as overwintering sites. Adults spend the

winter in attics, window wells, bedrooms, and between walls until spring, when they swarm out of the houses to breed and feed for the summer.

DCNR's Bureau of Forestry gets almost as many calls each year complaining about these big lady beetles as we do about gypsy moths and hemlock woolly adelgids. Our recommendation: vacuum them up and shake the bag outside. Also, seal up areas of your home that may allow them to get inside.

Chemical control Recent research has shown that an adelgid-killing chemical injected into tree boles or applied to the ground as a soil drench or soil injection may kill hemlock woolly adelgid, and prevent new established infestations for over a year. This has prompted PA DCNR to institute a chemical suppression program on public lands at 146 sites in the state. This type of control is restricted to large, high-value (ecological, historical, or aesthetic) trees. These trees may be too tall for biocontrol releases or for application of horticultural oils or soaps. Drawbacks of chemical control include high cost of treatment, temporary control, and secondary outbreaks of spider mites. It is our hope that chemical control can be used as a "stop gap" measure to stave the hemlock woolly adelgid off and give biological control time to take effect.

Silvicultural control The DCNR is attempting to restore areas that have been impacted by the hemlock woolly adelgid. This sometimes involves replanting with native species, such as eastern white pine, that are similar ecologically, but are not affected by hemlock woolly adelgid.

Host Resistance The DCNR hopes to increase our cooperation with researchers who are attempting to identify individual eastern hemlock trees that seem to be tolerant of hemlock woolly adelgid feeding. The seed source from these individuals could be used in regeneration programs. Other researchers are currently attempting to hybridize eastern hemlock with a more tolerant or resistant host. Unfortunately attempts to hybridize eastern hemlock with three Asian species have not been successful. Also, attempts to hybridize eastern hemlock with the

morphologically similar western hemlock or mountain hemlock have been unsuccessful because these species are not well adapted to the east coast climate.

What can landowners do about Hemlock Woolly Adelgid?

Chemical Control Options: What can be done depends on the value of the trees you wish to protect. Individual ornamental trees, small trees, or even several larger hemlocks in a landscape environment can be treated with insecticides. There are several spray materials registered for application to hemlocks by ground-spraying equipment and by injection techniques. Some sprays are relatively safe to the environment, such as horticultural oils and insecticidal soaps, and others are more toxic. Oils and soaps work by suffocating the adelgid. The best time to treat is either in spring and early summer when crawlers are present, or in fall when adults break dormancy. Sprays must completely drench the needles and twigs of the entire tree to be effective, therefore this method is only recommended for trees that are 30 feet in height or less. Applications of special insecticides can be made to the tree trunk or to the soil around the tree roots. This way the tree actually moves the chemicals up to the twigs and needles where the adelgids are feeding.

Some insecticides registered for control of hemlock woolly adelgid are labeled for homeowner use. Other insecticides are restricted for use only by licensed certified pesticide applicators. Check with your county cooperative extension agent or local pest management specialist for more information, and always **read and follow the pesticide label directions**.

Both commercial spraying and injection are expensive and results with either method vary greatly depending on the quality of the equipment used, the experience of the applicator, and treatment timing (adelgid development, wind, rain, soil moisture, etc.). Call several reputable tree service companies for pricing, ask for references, and be sure to check with local clients they have served.

Forest landowners with dozens or hundreds of infested hemlocks have very few options: inject, harvest, or wait-and-see. Most Pennsylvania landowners are watching and hoping that their hemlocks survive. Some are selling their commercial hemlock timber, knowing that dead hemlock degrades very rapidly.

Tips for maintaining hemlocks and avoiding or decreasing hemlock woolly adelgid infestations:

- Do not disturb shallow roots with heavy equipment or by digging or tilling;
- Keep hemlocks well-watered (apply about 1 inch / week around drip line) during droughts;
- Do not place a bird feeder amongst your hemlock trees in infested areas of the state. Birds can transport hemlock woolly adelgid crawlers to your trees.
- Remove large, heavily infested trees that can act as reservoirs for uninfested trees.
- Clip and burn heavily infested hemlock branches. If you can catch the infestation early enough, this may significantly slow the insect's spread and build-up.
- Do not change the grade (slope of the land) near hemlocks, such as excavations or tree wells;
- Do not change water runoff patterns around hemlocks. Simply moving a downspout or installing a patio can stress these trees;
- Do NOT fertilize trees infested with hemlock woolly adelgid with nitrogen. Researchers have found five times as many hemlock woolly adelgids on nitrogen-fertilized trees, regardless of whether fertilization occurred at infestation or six months later. Once an infestation has been eradicated, fertilize hemlocks lightly with a balanced fertilizer, such as 5-10-5, late in the fall.
- When applying lime or weed killers to lawn areas, keep them at least 10 feet away from the drip line (tips of outermost branches) of hemlock trees.

PEST ALERT

Walnut Twig Beetle and Thousand Cankers Disease of Black Walnut

Within the past decade an unusual decline of black walnut (*Juglans nigra*) has been observed in several western states. Initial symptoms involve a yellowing and thinning of the upper crown, which progresses to include death of progressively larger branches (Figure 1). During the final stages large areas of foliage may rapidly wilt. Trees often are killed within three years after initial symptoms are noted. Tree mortality is the result of attack by the walnut twig beetle (*Pityophthorus juglandis*) and subsequent canker development around beetle



Figure 1. Rapidly wilting black walnut in the final stage of thousand cankers disease.



Figure 2. Coalescing branch cankers produced by *Geosmithia*. Note the whitish sporulation of *Geosmithia* in lower left gallery.

development around beetle galleries caused by a fungal associate (*Geosmithia* sp.) of the beetle (Figure 2). A second fungus (*Fusarium solani*) is also associated with canker formation on the trunk and scaffold branches. The proposed name for this insect-disease complex is *thousand cankers*.

Walnut Twig Beetle

Distribution. The walnut twig beetle is native to North America, being originally described in 1928 based on specimens collected in the area of “Lone Mountain”, New Mexico (Lincoln County). In the 1992 catalog of Bark and Ambrosia Beetles by Wood and Bright the primary range of the insect was listed to include New Mexico, Arizona, and Chihuahua, Mexico (Figure 3). This range appears to coincide largely with the distribution of Arizona walnut (*J. major*), the likely original native host. In Arizona walnut the insect functions as a “typical” twig beetle, confining its development to overshadowed or injured small diameter limbs and functioning as a natural pruning agent.

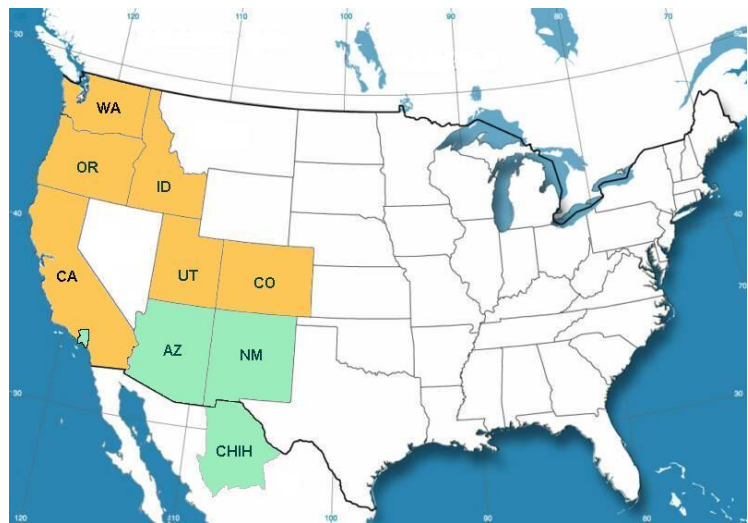


Figure 3. Distribution of the walnut twig beetle. In green are states and the California county of Los Angeles with records of the species prior to 1992. States in orange have reported the insect since 1998.

Isolated captures of the walnut twig beetle in California were first recorded in 1959 in Los Angeles County in association with both black walnut and southern California walnut, *J. californica*. Since 2002, it has been recovered extensively in the state and is thought to be established throughout most of southern California and the Central Valley. Northern California walnut, *J. hindsii*, has also been noted to be a suitable host for the beetle.

The first published record of a cluster of black walnut mortality associated with the walnut twig beetle was in the Espanola Valley of northern New Mexico where large numbers of mature black walnut died in 2001. However, this may have been preceded in Utah where undetermined black walnut mortality occurred in the early 1990s along the Wasatch Front; a Utah record of the beetle dates to 1988. Similar widespread decline also occurred about this time in the Boise-Meridian area of Idaho where the twig beetle was first confirmed present in 2003. Black walnut declines have been noted in some Front Range communities in Colorado since 2001 and the twig beetle has been confirmed present in the state since 2004. In those communities where the insect has been detected, the majority of black walnut has since died. *P. juglandis* has been recorded from Oregon (Portland) since 1997, has been commonly captured in funnel traps in The Dalles since 2004, and is suspected of being associated with recent widespread death of *Juglans* spp. in the Willamette Valley of Oregon. Walnut decline associated with the presence of walnut twig beetle were reported from the Prosser area of Washington in 2008.

Prior to these recent reports, walnut twig beetle was not associated with any significant *Juglans* mortality. In most areas where the die-offs of black walnut have occurred, drought was originally suspected as the cause of the decline and death of trees, with the beetle as a secondary pest. The widespread area across which *Juglans* spp. die-off have been recently reported, the documented presence of an associated canker-producing fungal pathogen carried by the twig beetle, and the occurrence of black walnut death in irrigated sites not sustaining drought, all suggest an alternate underlying cause.

Description. The walnut twig beetle *Pityophthorus juglandis* is a minute (1.5-1.9 mm) yellowish-brown bark beetle, about 3X long as it is wide. It is the only *Pityophthorus* species associated with *Juglans* but can be readily distinguished from other members of the genus by several physical features (Figures 4, 5). Among these are 4 to 6 concentric rows of asperities on the prothorax, usually broken and overlapping at the median line. The declivity at the end of the wing covers is steep, very shallowly bisulcate, and at the apex it is generally flattened with small granules.



Figure 4. Walnut twig beetle, side view. Photograph by Jim LaBonte, Oregon Department of Agriculture.



Figure 5. Walnut twig beetle, top view. Photograph by Jim LaBonte, Oregon Department of Agriculture.

Life History and Habits. Despite its small size - and pending common name - attacks by adult *P. juglandis* and larval development in black walnut rarely occur in twigs. Instead tunneling is almost always confined to branches greater than 2 cm diameter. Very large branches and even the trunk can be colonized.

Winter is spent primarily, and possibly exclusively, in the adult stage sheltered within cavities excavated in the bark of the trunk. Adults resume activity by late-April and most fly to branches to mate and initiate new tunnels for egg galleries; some may remain in the trunk and expand overwintering tunnels. During tunneling the *Geosmithia* fungus is introduced and subsequently grows in advance of the bark beetle (Figure 6). Larvae feed for 4-6 weeks under the bark in meandering tunnels that run perpendicular to the egg gallery (Figure 7) and pupate at the end of the tunnel.



Figure 6. Walnut twig beetle and associated staining around tunnel.



Figure 7. Walnut twig beetle tunneling under bark of large branch.

Adults emerge to produce a second generation. Peak flight activity of adults occurs from mid-July through late August and declines by early fall as the beetles enter hibernation sites. A small number of beetles produced from eggs laid late in the season may not complete development until November.

Cankers



Figure 8. Conidiophores and conidia of *Geosmithia*.

Two different types of cankers have been observed on declining walnut trees. Small, diffuse, dark brown to black cankers, caused by an unnamed fungus in the genus *Geosmithia*, initially develop around the tunnels of the walnut twig beetle (Figure 8). The fungus has also been cultured from beetles that emerge from black walnut.

Branch cankers may not be visible until the outer bark is shaved to expose the tunnels, although a dark amber stain may form on the bark surface in association with the cankers. Cankers expand rapidly and develop more expansively lengthwise than circumferentially along the stem. On thick barked branches and the trunk, canker may at first be localized in outer bark tissue and extend into the cambium only after extensive bark

discoloration has occurred. Eventually multiple cankers coalesce to produce girdling that results in branch dieback. The number of cankers that are formed on branches and the trunk is enormous; hence the name **thousand cankers** to describe the disease.

There appears to be a range in susceptibility of *Juglans* species to the *Geosmithia* fungus. Black walnut is a very susceptible host, with large cankers developing in response to inoculation. Other species, including Arizona walnut and little walnut (*J. microcarpa*) develop more restricted cankers following artificial inoculation. Evaluation of *Juglans* and related potential nut tree hosts (e.g., *Carya*) for susceptibility to thousand cankers disease will be determined in future studies.

A second canker type may occur on black walnut trees in advanced stages of decline. These diffuse cankers are much larger than branch cankers caused by *Geosmithia* and often exceed two meters in length, extend from the ground into the scaffold branches, and may encompass more than half the circumference of the trunk (Figure 9). Trunk cankers are not readily visible without removal of the outer bark. However, a dark brown to black stain on the bark surface or in bark cracks often indicates the presence of a canker. The inner bark and cambium below the bark surface on the canker face is macerated, water-soaked and stained dark brown to black. Both the walnut twig beetle and *Geosmithia* are found in the macerated bark but a second fungus *Fusarium solani* also has been consistently isolated from canker margins. The importance of *Fusarium solani* in the development of these trunk cankers is still being determined.



Figure 9. Large trunk cankers of black walnut associated with *Fusarium solani*.

Management

Controls for thousand cankers disease have not yet been identified and their development will require better understanding of the biology of the walnut twig beetle and the canker-producing *Geosmithia* fungus. Because of the extended period when adult beetles are active and the extensive areas of the trees that are colonized, foliar insecticide spray applications likely have limited effectiveness. Furthermore, colonization of the bark and cambium by *Geosmithia* may continue even if adult beetles or larvae are killed by the insecticide. The involvement of the fungus also will likely limit the ability of systemic insecticides to effectively move in plants in a manner to kill beetles. Rapid detection and removal of infected trees currently remains the primary means of managing thousand cankers disease. Drenching insecticides applied to the trunk in late summer may also be useful in killing beetles as they seek winter hibernation quarters.

For further information concerning the walnut twig beetle and the thousand cankers disease of walnut, contact Whitney Cranshaw (Whitney.Cranshaw@ColoState.EDU) or Ned Tisserat (Ned.Tisserat@ColoState.EDU), Department of Bioagricultural Sciences and Pest Management, Colorado State University.



Forest Health Factsheet

Gypsy Moth

Gypsy moth, *Lymantria dispar* (L.), is an important pest of hardwoods in the northeastern U.S. since its introduction from Europe to Massachusetts in 1869. It is now established in 19 states from Maine to Wisconsin and extends to northeastern North Carolina. In Pennsylvania it was first discovered in Luzerne and Lackawanna counties in 1932. A total of 4.3 million acres were defoliated in the state during the historical peak year of 1990. Suppression programs have been carried out by the Pennsylvania Bureau of Forestry since 1968 to minimize its impacts on the forests.

Hosts

Gypsy moth is a spring defoliator with more than 300 host species. Oaks, especially white oaks, are preferred by feeding caterpillars. Older larvae will also feed on conifers such as hemlock, pines, spruces and southern white cedar. Non-hosts include ash, yellow-poplar, sycamore, black walnut, catalpa, locust, American holly, and shrubs such as mountain laurel, rhododendron and arborvitae.



Mature larva

Life History

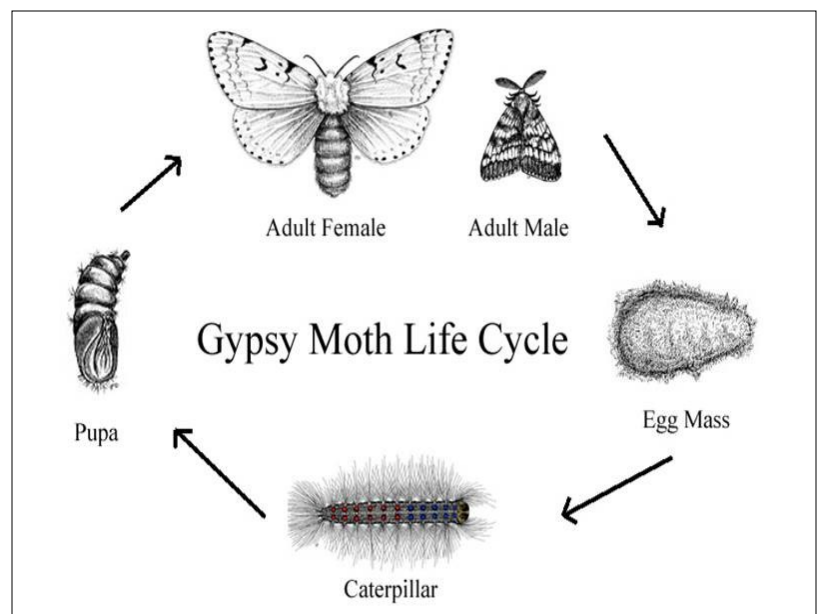
Gypsy moth has one generation per year in Pennsylvania. Females lay their eggs as light tan egg masses (400-600 eggs) on trees, stones and other substrates during July to overwinter. Eggs hatch from late April to early May the following spring. Small 1st instar larvae move in search of suitable food sources by floating in the wind on silken threads. Early instars feed on foliage and remain on hosts, whereas half-grown larvae usually feed in the canopy at night and move down from the tree to seek shelter in bark crevices and other protected sites during the day. Larvae mature by mid-June. Late instars are marked by five pairs of dark blue spots and six pairs of red spots along the back. Pupation takes place in late June or early July in places such as tree trunks, stone surfaces and building exteriors. Adults start to emerge two weeks later and reach peak emergence by mid-July.



Adult female and egg mass

Natural Enemies

Natural enemies (predators, parasitoids, and pathogens) play an important role in regulating gypsy moth populations. Birds, mammals, and predaceous insects such as the Calosoma beetle feed on eggs, larvae and adults. Parasitoids of gypsy moth include *Ooencyrtus kuvanae* for eggs, and *Cotesia melanoscelus* and *Parasetigena agilis* for larvae. There are also two pathogens that greatly affect gypsy moth caterpillars: the nucleopolyhedrosis virus (NPV), and the fungus *Entomophaga maimaiga*. Gypsy moth larvae die of viral infection hanging from trees in an inverted V position; while those killed by the fungus remain mummified head-down on the tree.



Management

The Pennsylvania Bureau of Forestry conducts annual egg mass surveys to monitor gypsy moth populations. A suppression program is planned when populations exceed threshold levels. Treatments are only conducted at the request of the landowner and if the area meets the program requirements. Treatment is applied when 50% of the caterpillars are in their second instar in the spring so timing is critical. If you believe that you have a need for a gypsy moth suppression treatment you should contact your gypsy moth county coordinator during the summer. Contact numbers and additional information on program requirements can be found at the [PA Bureau of Forestry Gypsy Moth Site](#).

Mechanical

Tactics for mechanical removal of gypsy moth egg masses can be effective for individual yard trees but are not effective as a forest-wide control method. Methods include removal of egg masses before they hatch and removal of objects where egg masses can be hidden by females. Another control tactic is wrapping burlap around the trunks of trees where gypsy moth larvae can hide during the day. The larvae hiding under the burlap are then scraped into a can of soapy water, killing the larvae. Sticky tape around the trees can also be used to entrap larvae as they move down the trees to hide during the day.



Burlap wrap

Insecticides

The principal insecticide used by the Pennsylvania Bureau of Forestry for gypsy moth suppression contains the bacteria *Bacillus thuringensis* var. *kurstaki* (Btk). This insecticide must be ingested by the early instar larvae and is more effective on the first three instars of gypsy moth. There are several chemical insecticides that can be used for gypsy moth control. Diflubenzuron is an insect growth regulator that must be ingested by the caterpillar and acts on the juvenile stages of invertebrates by preventing the formation of a new exoskeleton when the organism molts. It is effective against gypsy moth larvae. Another insecticide used in forestry applications is tebufenozide, an insect growth regulator which causes a premature molt in the caterpillars of butterflies and moths that feed on foliage treated with the insecticide. There is a biological insecticide containing the nucleopolyhedrosis virus registered under the name GYPCHEK. Since this virus specifically attacks gypsy moth, GYPCHEK is used in areas where rare and endangered butterflies and moths are believed to be present. This insecticide is produced in limited amounts by the USDA Forest Service.

References

1. [Woody Ornamental Insect, Mite, and Disease Management](#)

For More Information

[Gypsy Moth in North America](#)
[PSU Ext Gypsy Moth Factsheet](#)
[Gypsy Moth USDA FS Forest Insect and Disease Leaflet 162](#)
[Gypsy Moth in Wisconsin / Biological Controls](#)
[Homeowner's Guide to Gypsy Moth Management](#)

For more information contact:

Division of Forest Pest
Management @ 717-783-2066

<http://www.dcnr.state.pa.us/forestry/insectsdisease/index.htm>



pennsylvania
DEPARTMENT OF CONSERVATION
AND NATURAL RESOURCES

Pest Alert

Animal and Plant Health Inspection Service
Plant Protection and Quarantine

Spotted Lanternfly (*Lycorma delicatula*)

The spotted lanternfly is an invasive pest, primarily known to affect tree of heaven (*Ailanthus altissima*). It has been detected on many host plants, including apples, plums, cherries, peaches, nectarines, apricots, almonds, and pine. It also feeds on oak, walnut, poplar, and grapes. The insect will change hosts as it goes through its developmental stages. Nymphs feed on a wide range of plant species, while adults prefer to feed and lay eggs on tree of heaven (*A. altissima*).¹ If allowed to spread in the United States, this pest could seriously harm the country's grape, orchard, and logging industries.

Distribution and Spread

The spotted lanternfly is present in China, India, Japan, South Korea, and Vietnam. The insect was detected in Pennsylvania in September 2014. This was the first detection of spotted lanternfly in the United States.

Spotted lanternflies are invasive and can spread rapidly when introduced to new areas. While the insect can walk, jump, or fly short distances, its long-distance spread is facilitated by people who move infested material or items containing egg masses.

Damage

Both nymphs and adults of spotted lanternfly cause damage when they feed, sucking sap from stems and leaves. This can reduce photosynthesis, weaken the plant, and eventually contribute to the plant's death. In addition, feeding can cause the plant to ooze or weep,



Adult spotted lanternfly

resulting in a fermented odor, and the insects themselves excrete large amounts of fluid (honeydew). These fluids promote mold growth and attract other insects.

Description

Adult spotted lanternflies are approximately 1 inch long and one-half inch wide, and they have large and visually striking wings. Their forewings are light brown with black spots at the front and a speckled band at the rear. Their hind wings are scarlet with black spots at the front and white and black bars at the rear. Their abdomen is yellow with black bars. Nymphs in their early stages of

development appear black with white spots and turn to a red phase before becoming adults. Egg masses are yellowish-brown in color, covered with a gray, waxy coating prior to hatching.

Life Cycle

The spotted lanternfly lays its eggs on smooth host plant surfaces and on non-host material, such as bricks, stones, and dead plants. Eggs hatch in the spring and early summer, and nymphs begin feeding on a wide range of host plants by sucking sap from young stems and leaves. Adults appear in late July and tend to focus their feeding on tree of heaven (*A. altissima*) and grapevine

¹In Pennsylvania, adult spotted lanternflies have also been found feeding and egg laying on willow, maple, poplar, and sycamore, as well as on fruit trees, like plum, cherry, and peach.

(*Vitis vinifera*). As the adults feed, they excrete sticky, sugar-rich fluid similar to honeydew. The fluid can build up on plants and on the ground underneath infested plants, causing sooty mold to form.

Where To Look

Spotted lanternfly adults and nymphs frequently gather in large numbers on host plants. They are easiest to spot at dusk or at night as they migrate up and down the trunk of the plant. During the day, they tend to cluster near the base of the plant if there is adequate cover or in the canopy, making them more difficult to see. Egg masses can be found on smooth surfaces on the trunks of host plants and on other smooth surfaces, including brick, stone, and dead plants.

Report Your Findings

If you find an insect that you suspect is the spotted lanternfly, please contact your local Extension office or State Plant Regulatory Official to have the specimen identified properly.

To locate an Extension specialist near you, go to the U.S. Department of Agriculture (USDA) Web site at **www.nifa.usda.gov/Extension**. A directory of State Plant Regulatory Officials is available on the National Plant Board Web site at **www.nationalplantboard.org/membership**.



Nymphs are black with white spots in early stages of development. (Credit: itchydogimages)



Nymphs turn red just before becoming adults. (Credit: itchydogimages)



Hatched and unhatched egg masses



Cluster of adults on the trunk of a tree at night

Appendix D

Invasive Aquatic Species Identification Sheets

RUSTY CRAYFISH

Orconectes rusticus

The rusty crayfish is an aggressive crayfish with a huge appetite. It has not only managed to outcompete native crayfish species, but has also devastated aquatic ecosystems in several states including Pennsylvania.

SPECIES DESCRIPTION

Adult rusty crayfish are typically 7.5-13 cm (3-5 in) long with large, robust claws that display black banded tips. Coloration is typically a grayish-green. The best way to identify the rusty crayfish is by a set of dark rusty orange spots on each side of the carapace, which is its protective outer covering. The spots look as though the crayfish was picked up with a painted forefinger and thumb. Due to the hybridization of male rusty crayfish with female native crayfish, these spots may not always be present or well developed. Rusty crayfish also have smooth, instead of serrated mouth-parts.

Photo courtesy of
U.S. Geological Survey.

NATIVE & INTRODUCED RANGES

The rusty crayfish is native to the Ohio River basin. Its range extends through the states of Ohio, Kentucky, Tennessee, Illinois, and Indiana. In the early 1960s, the rusty crayfish was found outside its native range in a number of Wisconsin lakes and streams, and by 1967 was found in Minnesota streams. Today the rusty crayfish has spread to Michigan, Missouri, Iowa, New Mexico, New York, New Jersey, Pennsylvania, the New England states, and Ontario, Canada. Rusty crayfish were first discovered in Pennsylvania in 1976 in the lower Susquehanna River with established populations documented in the south central region; however, surveys are still needed throughout Pennsylvania to determine the true extent of its range.

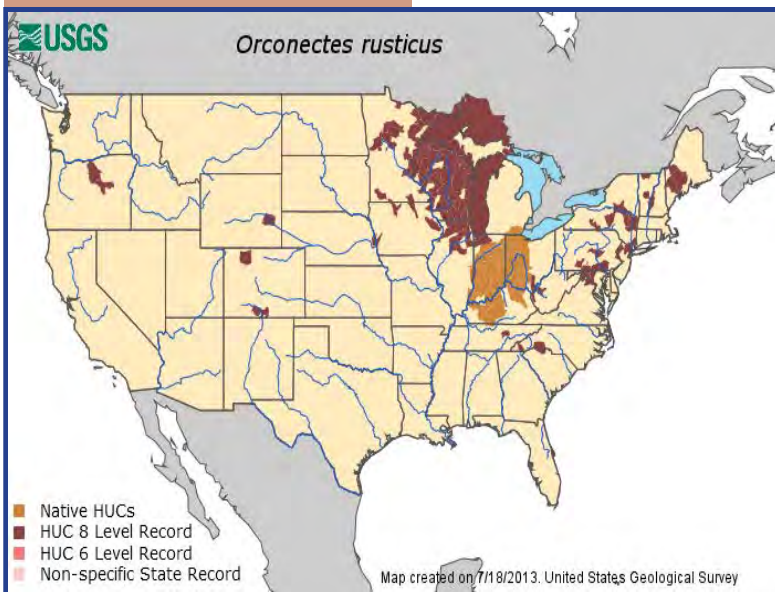
BIOLOGY & SPREAD

These invaders most likely spread by bait shops acquiring crayfish from out of state suppliers to use as bait for bass and trout. Anglers often then release the rusty crayfish after fishing. As a result, invasive populations of rusty crayfish have increased throughout the regions where they were sold.

They were also distributed by biological supply companies, which sold them to schools who may have released them into the wild.

HABITAT

Rusty crayfish can survive in a variety of habitats including lakes, rivers, ponds, and streams and prefer areas with adequate rock, log, and debris cover. They are often found in silt, clay, or gravel substrates, and are most active at temperatures above 46°F (8°C). Adult rusty crayfish mate in late summer, early fall, or spring. It is not necessary to have both a male and a female to establish a new invasion; a female carrying viable sperm could begin a new population if released into a suitable environment. The male transfers sperm to the female which she can store in her body until her eggs are ready to be fertilized, usually in the spring as water temperatures increase.



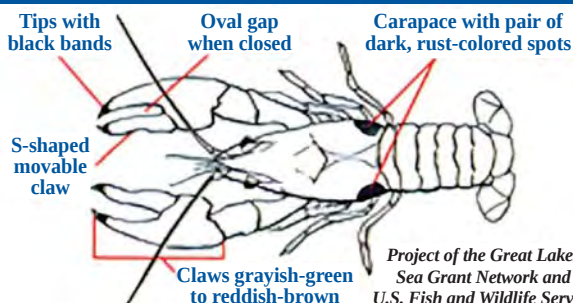
RUSTY
CRAYFISH

RUSTY CRAYFISH

Photo courtesy of U.S. Geological Survey.



How to Identify Rusty Crayfish



Project of the Great Lakes
Sea Grant Network and
U.S. Fish and Wildlife Service



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IMPACTS

Threat to Biodiversity

The rusty crayfish is very aggressive and often displaces native crayfish species. Studies show that rusty crayfish can eat twice as much as similar-sized native crayfish, consuming plants, aquatic worms, snails, leeches, clams, aquatic insects and crustaceans, decaying plants and animals, fish eggs, and small fish. Their food choices make them competitors with native crayfish as well as small fish. Rusty crayfish grow rapidly and quickly become too large for fish to eat, removing themselves from a critical part of the food chain. They also impact native crayfish reproduction as male rusty crayfish will mate with female native crayfish. This produces sterile offspring and reduces native crayfish populations. Perhaps the most serious impact of the rusty crayfish is destruction of aquatic plant beds. Rusty crayfish devour so much underwater vegetation that food, shelter, and spawning sites for other organisms are dramatically reduced.

Economic Costs

By destroying aquatic plant beds and affecting habitat, the presence of the rusty crayfish can result in reductions in fish production, including valued sport fish. In heavily infested lakes such as in Wisconsin and Minnesota, recreational swimming has declined due to rusty crayfish occupying favorite swimming areas during the day. Other crayfish species, even if abundant in these areas, do not deter swimmers because they are less conspicuous in daylight hours.

PREVENTION & CONTROL

Many chemicals kill crayfish; however, none are currently registered for crayfish control and none selectively kill rusty crayfish without killing other crayfish species. The best way to prevent further ecological problems caused by the rusty crayfish is to prevent or slow their spread into new waters. Become knowledgeable about the crayfish species in your area and learn how to identify them. Never release live bait into any water body, and never transport any crayfish from one water body to another. It is illegal to possess, sell, barter or transport rusty crayfish in Pennsylvania.

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GOLDFISH

Carassius auratus



Photo by
 Jon Sullivan,
 biolib.cz, Encyclopedia of Life

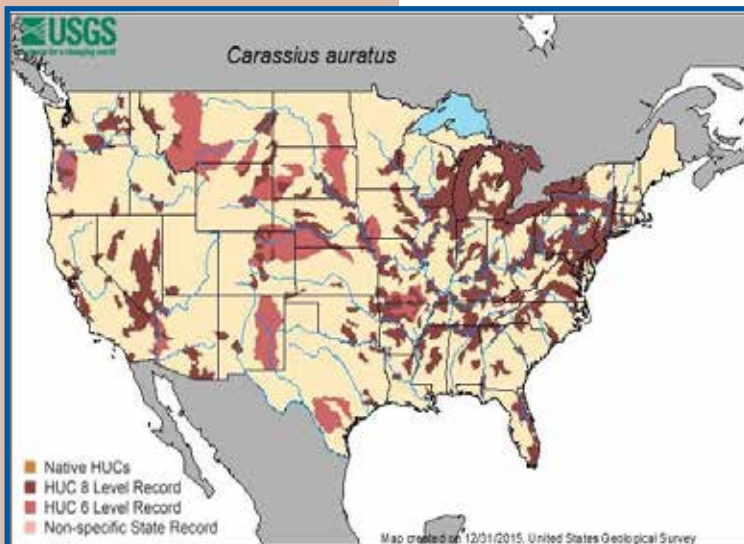
The goldfish is a member of the carp and minnow family. It was one of the first aquatic invasive species to reach North America, arriving in the 1600s as an ornamental fish for aquariums and water gardens. It is now one of the world's most widespread invasive species.

SPECIES DESCRIPTION

Goldfish have elongated, stout bodies which are typically 10-20 cm (4-8 in) in length and weigh 100-300 g (3.5-10.5 oz), although they can reach a maximum length of 59 cm (23 in) and a maximum weight of 6.6 lbs. They have a long dorsal fin with 15-24 rays, and a hard serrate spine at the origin of both the dorsal and anal fins. They typically have 26-32 scales on the lateral line. The mouth is small, lacks barbels and is situated in the middle of the head with both jaws the same length. While goldfish were mostly golden in color one thousand years ago, they now come in a variety of colors, including orange, yellow, white, black, silver, olive-green, or greenish-brown and combinations of these colors. When found in nature, goldfish are most often a shade of green, brown or gray.

NATIVE & INTRODUCED RANGES

Native to eastern Asia, goldfish have been reported invasive in the United States by every state except for Alaska. They are established in all of the Great Lakes, and in Erie, Northampton, and Philadelphia counties in Pennsylvania. Individual specimens have also been collected in several drainages throughout Pennsylvania; however, it is unknown whether these represent established populations.



National Range Map:
 USGS NAS Database

BIOLOGY & SPREAD

Goldfish have been intentionally introduced for ornamental purposes to ponds, fountains, and small lakes from which they may disperse through connecting waters. Many introductions of goldfish were also due to their use as live bait. In addition, goldfish are often released into the wild by pet owners not realizing the environmental repercussions of setting the fish free.

Photo courtesy of Pennsylvania Sea Grant



GOLDFISH



Photo courtesy of
Miroslav Fiala, Biolib.cz



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Management Program, The U.S. Fish and Wildlife
Service, and the Great Lakes Restoration Initiative

HABITAT

While goldfish prefer a habitat with a muddy bottom and thick vegetation, they can tolerate pollution, temperature fluctuations, and high levels of turbidity. They naturally live in freshwater ponds and slow-moving or still waters in depths of up to 19 m (65 ft) and prefer temperatures of 4-41°C (40-106°F), although they can not live for long at high temperatures.

IMPACTS

Threat to biodiversity

Goldfish are believed to be responsible for population declines in many native fish, invertebrate, and plant species. They feed on snails, small insects, fish eggs, and young fish, making it a competitor with and a predator of native fish. They also stir up mud and other matter when they feed, increasing the cloudiness of the water and uprooting native plants. They are also very prolific and can produce very large populations.

Health risks

Goldfish have the potential to carry diseases such as koi herpesvirus that can harm local fish populations.

PREVENTION & CONTROL

Preventing the introduction and spread of this species into water bodies is the best and most cost-effective way to control goldfish. Never release unwanted aquarium pets and plants into the wild. Instead look for alternatives, such as contacting a retailer for proper-handling advice or for possible returns. Give or trade with another aquarist, pond owner, or water gardener, or donate to a local aquarium society, school, or aquatic business. Disposal of live organisms should be considered a last resort; however, if this is an option be sure to contact a veterinarian or pet retailer for guidance about humane disposal of animals.

If prevention efforts fail and goldfish are introduced to a new location, know how to identify and report them. Always check for and remove any plants, mud, and debris from boats, trailers, clothing, and equipment before leaving a water body. Clean gear and equipment with either hot water (104°F or 40°C) or salt water and let dry thoroughly for five days before entering a new water body. Since young goldfish can resemble baitfish, it is important to drain water from bait buckets, bilges, and live wells before transporting to new areas.



Photo courtesy of
Biopix, Encyclopedia of Life

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AIS

Aquatic Invasive Species

CHINESE MYSTERY SNAIL



Photo from "Kemongsa Science In Picture" Volume 21, published by Kemongsa, 1993.

COMMON NAME: Chinese Mystery Snail

Many of the Oriental mystery snail's common names are interchanged, Japanese mystery snail, Japanese Black snail and Japanese trapdoor snail. Some believe that they have identified populations of Japanese mystery snails in the United States but they may just be Chinese mystery snails misidentified.

SCIENTIFIC NAME: *Cipangopaludina chinensis*

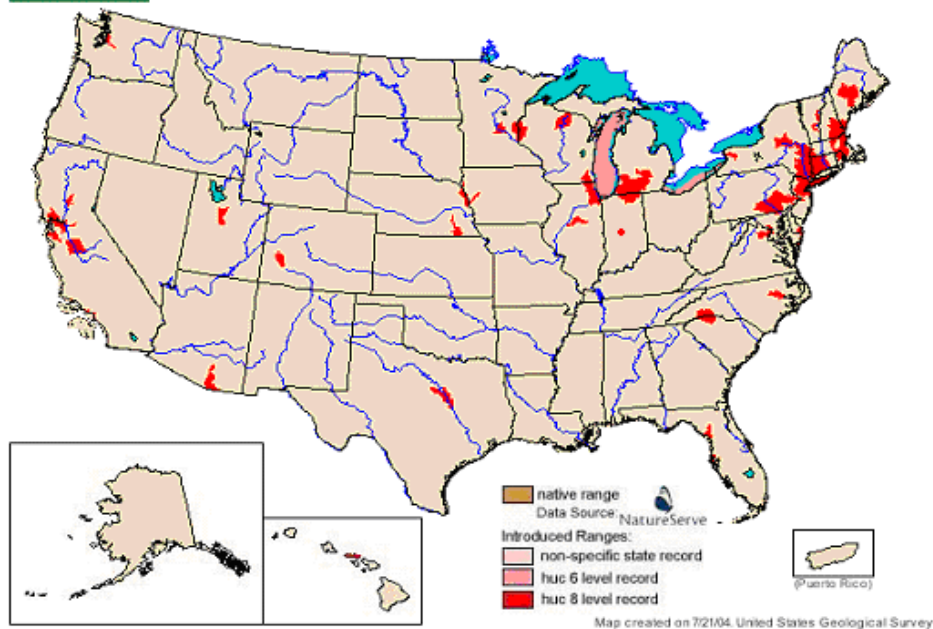
Other scientific names that will sometimes appear in older literature are *Viviparus chinensis malleatus*, *Viviparus japonicus*, *Viviparus stelmaphora*, *Paludina malleata*, *Paludina japonicus*, *Cipangopaludina malleata*, and the most common being *Viviparus malleatus*. *Cipangopaludina chinensis* has two different variations, var. *chinensis* and var. *malleata*. The Chinese mystery snail is in the mystery snail family, Viviparidae.

DISTRIBUTION: The Chinese mystery snail is native to Burma, Thailand, South Vietnam, China, Korea, Japan, the Philippines, and Java. It has been introduced into the United States in approximately 27 states. The Great Lakes have not gone unaffected by the Chinese mystery snail, Lake Michigan and Lake Erie have had introductions.

Indiana: Chinese mystery snails have been found in Fall Creek and the West Fork of White River, both locations are in Marion County.



Cipangopaludina chinensis



DESCRIPTION: The shell of the Chinese mystery snail is smooth and strong. It is a uniform color throughout without banding and is usually a light to dark olive-green. The shell can have 6 to 7 whorls. The whorls are strongly convex and each suture is very indented. The outer lip is either round or oval and has a black color to it. The shell can grow to a couple of inches in length.

LIFE CYCLE BIOLOGY AND LIFE HISTORY: You can find Chinese mystery snails in lakes, ponds, rice paddies, irrigation ditches, roadside ditches, and the slower portions of streams where there is some sort of mud substrate. They will stay partially buried in the mud where the water is quiet. The females will give birth to live, crawling young. They eat zooplankton and phytoplankton. Mystery snails have a feature called a “trap door” which allows them to close up the opening in their shell when water conditions are unfavorable. This poses problems when trying to eradicate with chemicals because the Chinese mystery snail can close up and wait until the chemical has dissipated before opening again.

PATHWAYS/HISTORY: In 1892, Chinese mystery snails were imported into live markets in San Francisco. In 1911, a thriving population was found in the San Francisco Bay. They were found in Boston, Massachusetts in 1915 and in 1950 Florida reported finding a population. By 1965, Chinese mystery snails were established both on the west coast and on the east coast as well as some of the Gulf States like Texas. The great Lakes have been affected also; Lake Michigan and Lake Erie populations were reported in 1965.

DISPERSAL/SPREAD: Chinese mystery snails were probably introduced into the United States through releases from the aquarium industry. This snail is readily imported for Asian food markets. Therefore, some releases may have been intentional in an effort to create a local food source. Once in a body of water the Chinese mystery snail could be transported via bait buckets and water holding areas on boats.

RISKS/IMPACTS: Chinese mystery snails can serve as vectors for the transmission of parasites and diseases. Some of the parasites and diseases that the Chinese mystery snail has been known to play host to can infect humans. Their shells will clog the screens of water intake pipes inhibiting the flow of water. Also, they naturally compete with our native snails for food and space.

MANAGEMENT/PREVENTION: Specific control methods for the Chinese mystery snail have yet to be developed but there are some general snail management techniques that could be applied. Biological control is always a method that most people support because it usually causes the least amount of damage to other aquatic organisms. By introducing fish or turtles that eat snails you may be able to lower the population. There is also the option to use a chemical control method. There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With Chinese mystery snails possessing the ability to “close up”, more damage would probably occur to native snails in the treatment area than to the target pest.

The best type of control is prevention. Preventing any further spread of the Chinese mystery snail will help keep our native ecosystems healthy. To help stop the spread of the Chinese mystery snail a few simple steps should be followed.

- ✓ Learn to identify the Chinese mystery snail and other exotic snails.
- ✓ If you have snails or other animals in an aquarium and you no longer wish to care for them, you should euthanize the animals before disposing in the trash. **DO NOT RELEASE THEM IN THE WILD!**
- ✓ Remove mud, plants, fish, and animals from all of your equipment and drain all water from the bilge and livewells before leaving the launch area.
- ✓ Clean your equipment with hot water or a pressure washer and allow it to dry for five days before transporting it into a new body of water.
- ✓ Never release plants or animals into a different body of water from which they came.

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Updated 3/05



ASIAN CLAM

Corbicula fluminea

Photo courtesy of Noel M. Burkhead,
United States Geological Survey NAS.



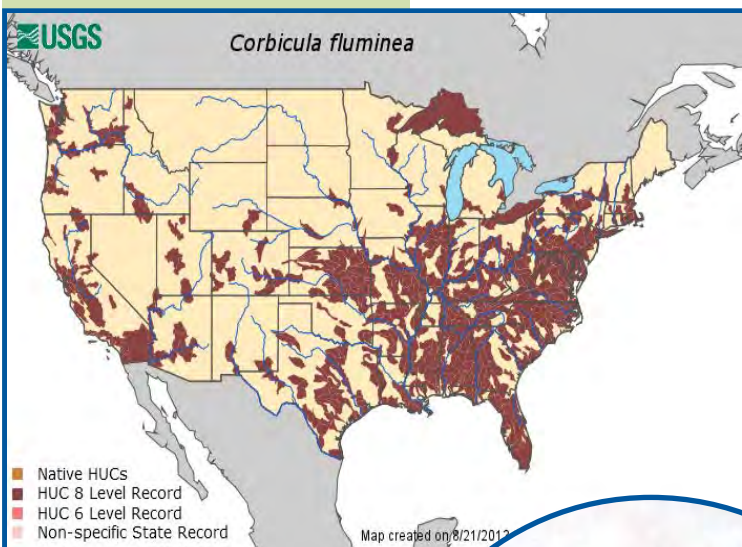
ASIAN CLAM

The Asian clam, also called the Asiatic clam, pygmy clam, or gold clam, is a small, freshwater bivalve that rarely exceeds the size of a nickel. This biofouler has spread rapidly in lakes, canals, streams, rivers, and reservoirs, threatening ecosystems and lakeside communities all throughout North America.

SPECIES DESCRIPTION

The shell of the Asian clam is thick, triangular in shape, and has deep distinctive growth rings. They are typically small, averaging less than 2.5 cm (1 in) and rarely exceeding 6.5 cm (2.5 in). There are two morphs of Asian clam in the United States. A lighter morph, with an outside shell typically yellow-green to brown in color is found in the northeast; and a darker morph exists in the southwestern United States. The inside of the shell is layered with polished, light purple nacre, and finely serrated teeth. Juveniles are free-floating and microscopic, called veligers, and appear under a microscope in a “D” shape less than 1 mm in length.

NATIVE & INTRODUCED RANGES



Map courtesy of United States
Geological Survey.

Native to the temperate and tropical regions of Southeast Asia and Africa, it is believed that the Asian clam was first introduced to the west coast of the United States in 1924 to be harvested as a food source. By the 1970s, it occupied most of the Mississippi River Basin, the Gulf Coast, and the eastern United States. In Pennsylvania, the Asian clam is widespread and can be found in the Allegheny, Monongahela, Ohio, Susquehanna, Delaware, and Schuylkill rivers and their tributaries as well as many other lakes and streams in Pennsylvania.

BIOLOGY & SPREAD

The Asian clam is a prolific and highly competitive species that is capable of rapid growth and spread. Because it is hermaphroditic, the Asian clam is capable of self-fertilization, and a single clam can release hundreds to thousands of free-floating, microscopic larva per day. In warmer waters this spawning activity can take place year round, creating ample opportunities for juveniles to be spread by water currents and human activity as they go unseen in bait buckets and livewells. Asian clams can be spread as they attach to boating, fishing, and scuba diving equipment and be transported to new waterways. They are also still sold commercially as bait throughout the United States, and in the aquarium trade as pygmy or golden clams.



Photo courtesy of Anna McCartney,
EDeBolt LGA.

ASIAN
CLAM

ASIAN CLAM



Photo courtesy of Randall Schietzelt,
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HABITAT

While Asian clams are a hardy and persistent species, they prefer running water with a sandy or gravel substrate bottom. Therefore, they are often found in streams, rivers, ponds, lakes, and constructed canals. They are considered a freshwater species, but can withstand slightly brackish waters and estuarine populations have been reported on both the east and west coasts. They also prefer to colonize in warmer areas near the shore with plenty of sunlight, and although they are able to withstand freezing conditions, their ability to reproduce decreases with exposure to lower temperatures.



Photo courtesy of Anna McCartney,
EDeBolt LGA.

IMPACTS

Threat to Biodiversity

Dense clusters of Asian clams, often reaching over 6,000 individuals/m², can alter the natural benthic community of an ecosystem. They are efficient filter feeders, consuming microscopic plants and animals from the base of the food chain and competing with native mussels, juvenile fish, and other native aquatic organisms for food. Since they are capable of tolerating polluted environments better than many native bivalves, Asian clams are displacing native species. In many areas, native mussels are in danger of becoming extinct due to the infestations of Asian clams.

Economic Costs

The Asian clam is a known biofouler, causing millions of dollars in damage each year for power plants and industrial water systems. Asian clams clog pipes, irrigation canals, and intake systems, and damage equipment like boat motors, diving gear, and commercial water systems. In addition, they impair the aesthetic and recreational values of public beaches, lake front properties, and swimming areas.

PREVENTION & CONTROL

Early detection is the first line of defense in preventing the spread of this species to new areas. Know how to identify the Asian clam and distinguish it from native mussels.

Always check for and remove plants, mud, and debris from boats, trailers, clothing, and equipment before leaving a water body. Drain all water from bait buckets, bilges, and live wells before transporting to new areas. Clean all gear and equipment with hot water or salt water, OR let boats and equipment dry thoroughly for at least five days before entering a new water body.

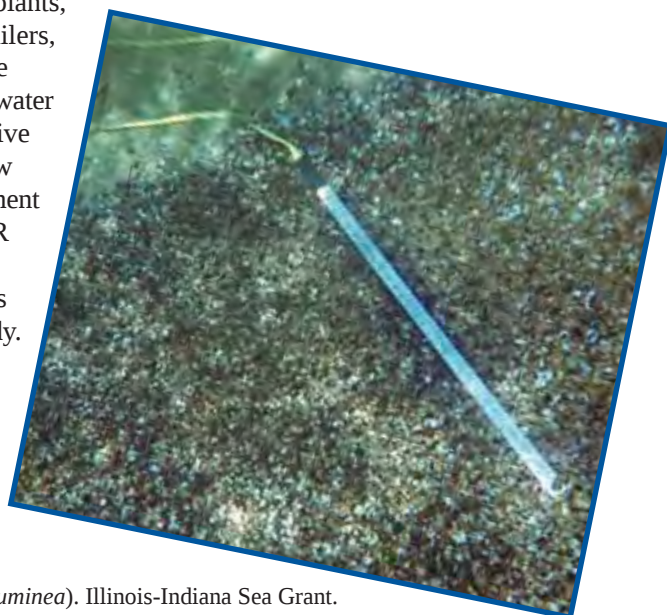


Photo courtesy of Anna McCartney,
EDeBolt LGA.

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AIS

Aquatic Invasive Species

COMMON CARP



COMMON NAME: Common Carp

Some varieties of common carp are called mirror carp, leather carp, koi, and Israeli carp.

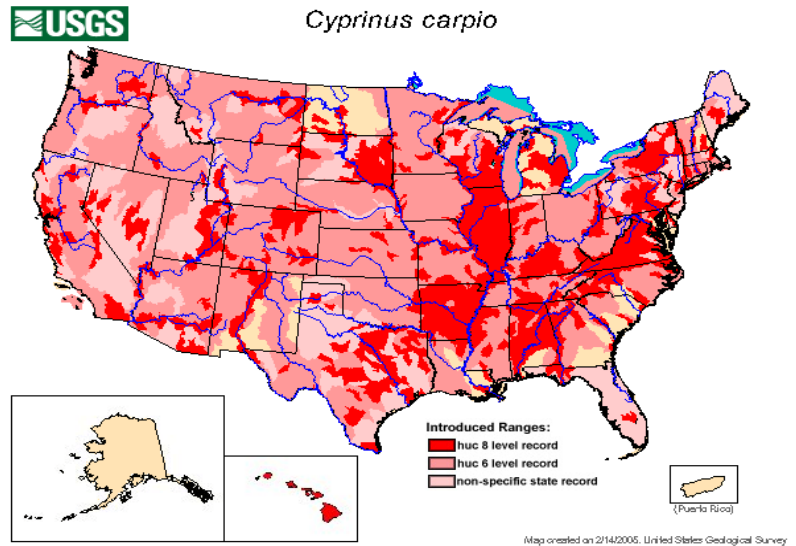
SCIENTIFIC NAME: *Cyprinus carpio*

Cyprinus is Greek and *carpio* is Latin; both words mean carp.

DESCRIPTION: Common carp is in the family Cyprinidae (minnow and carp family). *Cyprinus carpio* is easily identified by two pairs of barbels on each side of the upper jaw. These bronze, brassy or yellow fish have serrated dorsal and anal fin spines. Common carp is one of the largest members of the minnow family. Most of these heavy bodied fish are from 1 to 10 pounds and 12-25 inches in length. The world record catch for common carp is from Romania; that fish weighed just over 83 pounds! The Indiana state record common carp is 43 pounds and 4 ounces.

There are many varieties of common carp found throughout the world. Carp that are partially scaled along their sides are called mirror carp. Some common carp have few to no scales and are termed leather carp. Koi is a fancy breed of the common carp that are popular in small ponds and water gardens. While many people believe that the goldfish is a young carp, goldfish and common carp are actually two distinct species.

DISTRIBUTION: Common carp are distributed throughout the continental U.S. and range from central Canada to central Mexico. This species is one of the most widely distributed fish species in North America. In Indiana, common carp are found throughout the rivers and streams of the state, many natural lakes and impoundments, and some farm ponds. Carp can tolerate a variety of environmental conditions and habitat types which has allowed them to invade such a large geographical area.



LIFE CYCLE BIOLOGY: Common carp is a warm water species and does well in muddy, eutrophic (highly productive, rich in mineral and organic nutrients) waters. Peak spawning occurs from May through July in shallow waters. The sticky eggs (100,000 to 500,000 in number) are deposited on submerged vegetation and hatch in less than a week. Common carp fry feed on plankton. Juveniles and adults are found in deeper waters feeding predominantly on aquatic plants, algae and small invertebrates near the bottom.

PATHWAYS/HISTORY: *Cyprinus carpio* appears to have evolved in the Caspian Sea in western Asia. They then migrated to the Aral and Black Seas, east to mainland Asia and west as far as the Danube River. There is some controversy as to when common carp were first introduced into the U.S. Some suggest they were introduced in the early 1830's into New York while others report the first carp were brought into California in the early 1870's. The U.S. Fish Commission imported common carp from Germany in 1877 to establish a potential food source. From the early 1880s, this species was stocked in farm ponds and frequently escaped into open waters by means of floods and dam breaks. By 1885, the U.S. Fish Commission was stocking lakes and rivers throughout the United States.

DISPERSAL/SPREAD: Once established in a body of water, common carp can escape from the point of introduction and move to other connected bodies of water. Transfer of the species to different water bodies can also occur by anglers using juvenile carp as bait fish.

RISKS/IMPACTS: Although this species was originally introduced in the U.S. as a food source, it has yet to be widely accepted for this purpose. The bottom feeding habits (rooting) of this fish prove to be quite destructive. When overabundant, carp cause an increase in water turbidity and a decrease in aquatic plants and invertebrates. Evidence has also proven that the common carp prey on the eggs of other fishes and their foraging

activities can destroy spawning beds of more desirable species. Therefore, common carp are responsible for the decline of some native fish species.

Predators will feed on young carp. However, due to the fast growth of juvenile carp, they are vulnerable to predation for a short period of time. If a body of water is productive and predator populations are relatively low, common carp can dominate a fishery in a short period of time.

MANAGEMENT/PREVENTION: Physical removal methods such as seining, electrofishing, netting, and angling have proven ineffective at removing a large proportion of a carp population. Once an overabundant population is established, the most effective means of seriously reducing or eliminating a population is with complete lake drainage or the use of a fish toxicant. One must be certain that all upstream areas that harbor carp are also managed to eliminate this species, otherwise the downstream body of water that was either drained or the fishery eradicated will be at risk of a quick rebound of the species. In areas where carp have not reached a nuisance level, a dense predator base should be maintained to provide a high level of predation on young carp.

While carp are found throughout much of Indiana, there are bodies of water that do not contain this species. There are some things you can do to prevent spreading common carp to bodies of water not yet infested.

- ✓ Dispose of unused bait in the trash rather than in the water.
- ✓ Never transfer live fish from one body of water to another.
- ✓ It is illegal to use live common carp as bait in Indiana (312 IAC 9-6-8).

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AIS

Aquatic Invasive Species

ZEBRA MUSSEL



COMMON NAME: Zebra Mussel

The zebra mussel gets its name from the dark and light stripes on its shell that resembles those on a zebra.

SCIENTIFIC NAME: *Dreissena polymorpha*

Zebra mussels are in the Dreissenidae family, the false mussel and zebra mussel family.

DISTRIBUTION: Natively the zebra mussel inhabits parts of western Russia near the Caspian Sea and the Ural River. From its native origin, the species has spread to the point where the zebra mussel now affects the waters of most of Europe. The Canadian provinces of Quebec and Ontario have confirmed populations. As of 2005, sightings have been received from the following states: Alabama, Arkansas, Connecticut, Illinois, Indiana, Iowa, Kansas, Kentucky, Louisiana, Michigan, Minnesota, Mississippi, Missouri, Nebraska, New York, Ohio, Oklahoma, Pennsylvania, Tennessee, Vermont, Virginia, West Virginia, and Wisconsin. For the latest distribution of zebra mussels in the United States, please visit the following website:

http://nas.er.usgs.gov/taxgroup/mollusks/zebramussel/maps/current_zm_map.jpg

Indiana: To view a list of the known bodies of water in Indiana that contain zebra mussels, please visit:

http://www.in.gov/dnr/invasivespecies/zebra_mussels_sightings.pdf

DESCRIPTION: Zebra mussels have a triangular shaped shell that rarely exceeds 1.5 inches in length. Their shell is bivalve meaning it has two halves. Usually the shell will have alternating dark and light bands resembling the stripes of a zebra, hence their name. However, not all zebra mussels will have this characteristic coloring pattern, some may

be entirely dark or light. The most distinguishing characteristic to look for would be the tuft of fibers called the byssal threads that grow from the foot and through the hinge of the mussel. These threads allow the mussel to attach to any hard surface. A similar species that may be confused with the zebra mussel is the quagga mussel, another exotic species.



LIFE CYCLE BIOLOGY: Zebra mussels are able to reproduce their second year. Over one spawning season a single female zebra mussel is capable of releasing one million eggs. The eggs will be fertilized when the male and female expel their gametes at the same time. Fertilized eggs develop into larvae called veligers. These veligers are not visible to the naked eye and are about the width of a human hair. They can remain suspended in the water for 3 to 4 weeks before they find a hard surface to attach to; those that don't find a substrate to attach will die. Once attached it takes one year for it to grow one inch and become sexually mature. The maximum age reported in the U.S. is 3 years while in Europe they have been reported to live 4 to 6 years.

Their ability to attach to hard surfaces is due to the tuft of fibers located at the hinge of their shell called byssal threads. These threads produce a powerful glue that anchors the mussel in place. Any hard surface is a suitable place for a mussel to live such as rock, metal, wood, vinyl, glass, rubber, fiberglass, paper, plants, other mussels, and the bodies of slow moving animals like crustaceans and turtles. Zebra mussels will layer over each other forming a dense covering over the substrate. Juveniles have the ability to break their attachment and generate new threads which allows them to drift downstream and find a new home.

Zebra mussels are filter feeders and can filter one liter of water per day. Almost all matter in the water is filtered. Zebra mussels feed on phytoplankton and some small zooplankton. Other matter filtered is expelled as pseudofeces. The zebra mussel has some limiting factors such as water temperature, calcium, pH, substrate, salinity and nutrients. Zebra mussels can only tolerate temperatures from 32 °F to 96°F. They need a temperature of at least 54°F. to reproduce. A calcium level of more than 20 parts per million is necessary in order to survive. The zebra mussel needs an alkaline environment as well with a pH from 7.2 to 9.0. They do best in water where there is an abundance of hard substrate, but they can proliferate in soft sediments. They like freshwater environments with a salinity level of less than 4 parts per thousand. Zebra mussels also do not do well in water that has a high nutrient content.

PATHWAYS/HISTORY: From its native range in the Caspian Sea, the zebra mussel invaded Eastern Europe through a series of shipping canals that were built in the late 1700's and early 1800's. Most of Europe was invaded by the mollusk by 1830. The first zebra mussel sighting in the United States was in 1988 in Lake St. Clair. This lake connects Lake Huron to Lake Erie.

DISPERSAL/SPREAD: It is believed that the zebra mussel arrived in Lake St. Clair via the ballast water of transoceanic ships. It did not take long for the zebra mussel to spread. By 1990, they could be found in all of the Great Lakes. In 1991, zebra mussels

had found their way into the Illinois and Hudson Rivers. From here they had even more access to other rivers and to disperse ever further. Just one year later, established populations were found in the Arkansas, Cumberland, Hudson, Illinois, Mississippi, Ohio and Tennessee Rivers. It was only a matter of time before the zebra mussel made its way into the inland waters of some states. Populations are now known from at least 23 states, primarily within the Great Lakes and Mississippi River watersheds. To see a progression of the invasion of the mollusk in the United States you may visit the following website: http://cars.er.usgs.gov/Nonindigenous_Species/ZM_Progression/zm_progression.html

Both the movement of adult zebra mussels and the larval form, known as veligers, can infect other bodies of water. If an adult zebra mussel attaches to a boat it has a free ride to new water by individuals who trailer their boats from one body of water to another. Adult zebra mussels are able to close their shell and survive for several days in cool, moist conditions. In its veliger stage of life a zebra mussel is able to hitchhike to other lakes and streams in water being held in the bilge, live wells, or bait buckets, or they may cling to plant fragments, the boat or trailer, or any other equipment or recreational items coming into contact with water. Once in a new body of water, their prolific breeding allows them to easily establish a viable population. Once a population is established the downstream waters are at risk of infestation since veligers can float downstream for 3 to 4 weeks in search of a hard object to settle on.

RISKS/IMPACTS: Ecologically, the zebra mussels cause many problems. One such problem is that they need to attach to a hard surface to survive, and these hard surfaces could be anything from many manmade objects to other animals. Zebra mussels will attach to crayfish, turtle shells as well as other mussels. When a native mussel has zebra mussels attached, the native mussel loses its ability to move, feed, breath, and breed. Eventually this will lead to the death of the native mussel. In



Lake St. Clair and Lake Erie, the native mussel populations have been severely reduced due to the dense populations of zebra mussels. This dramatic drop happened only two years after the zebra mussel was discovered in the Great Lakes. This sends up a red flag for Indiana managers. With many of Indiana's native mussels already on state or federal endangered and threatened species list, zebra mussels could spell eventual extinction.

Zebra mussels have the ability to filter up to 1 liter of water per day. They eat the phytoplankton that is suspended in the water, which is in competition with the nearly microscopic animals called zooplankton. The food chain is very delicate and the zebra mussels could be disrupting it by taking out the very bottom link, the phytoplankton. This affects all the higher organisms including the fishes

Because zebra mussels filter large amounts of water, infested lakes have become clearer. While this may sound like a good thing, this can cause problems as well. With clearer water, sunlight penetrates to deeper water; this allows for more vegetative growth. This vegetation can become so thick that it could hinder swimming and boating.

While zebra mussels feed on phytoplankton, they do not eat blue-green algae. Because the blue-green algae are not being eaten, there is a competitive advantage over other algae resulting in a blue-green algae bloom. Such blooms occur in Lake Huron and Lake Erie much more frequently than ever before. Some forms of blue-green algae produce toxins. If enough blue-green algae toxin is produced, harm can occur to fish, waterfowl and any other animals that drink the water, including humans.

Yet another ecological impact that the zebra mussels impose on our native fauna is the fact that they are bioaccumulators. Any contaminant in the water is multiplied up to 10 times the water concentration in the zebra mussels. When the mussels are eaten, the contaminant concentration increases through the food chain. As zebra mussels become more prevalent more animals will eat them increasing the chance that higher levels of toxins will be accumulated throughout the food chain.

The zebra mussel is also economically detrimental. The mussel's need to attach to hard surfaces creates problems. Water intake structures are prime locations for attachment. These intakes provide a continuous flow of water and protection from predators. Once inside a pipe the zebra mussels layer on top of each other eventually causing reductions in pumping capabilities and even complete blockages. Industrial water pipes are not the only ones at risk. Residents whose cottages rely on lake water could see clogging of their supply lines. These pesky mussels get into engine cooling systems on boats, increase the deterioration of piers, increase the corrosion of steel and concrete, they have even sunk navigational buoys because the buoy could not support the additional weight. Beaches can become covered in the sharp zebra mussel shells and the odor given off by decaying mussels can be unpleasant, both of which result in reduced recreation. There is not just one problem associated with zebra mussels, as you can see they create a complex web of problems that are not easily fixed.

MANAGEMENT/PREVENTION: Once a population of zebra mussels has become established it is impossible to eradicate them without complete destruction of everything else that also lives in the water.

Research is ongoing in an attempt to find a way to disrupt the reproductive cycle of zebra mussels. Researchers are looking to try to mimic male hormones that trigger females to release their eggs. If successful, they can disrupt the breeding of the mussels by having the males and females release their gametes at different times, preventing fertilization.

Introducing natural predators, parasites, or diseases of the zebra mussel into an infested body of water is another way of lowering a population. This is known as biological control. Native Indiana predators of zebra mussels include waterfowl, sturgeon, yellow perch, freshwater drum, catfish, and sunfish. The exotic round goby, present in all of the Great Lakes and some of its tributaries, also has quite a liking for zebra mussels. One laboratory study observed one round goby eating 78 zebra mussels in a day. Unfortunately, anything that feeds heavily on zebra mussels will build up high levels of contaminants in its body. Due to the high reproductive capacity of zebra mussels, it is unlikely that predation will have a profound effect on reducing the nuisance caused by the invasive mussels.

There have been some successful procedures developed to prevent the invasive mussels from clogging water intakes. These include using molluscicides to kill mussels

at the entrance of water intake pipes, physically removing the mussels by scrapping, pigging, or high pressure washing, hot water or steam injection into infested pipes, and using toxic coatings containing copper or zinc on screens of intake pipes and boat bottoms to discourage attachment.

Many states have regulations prohibiting the import, transport, or possession of this species in order to limit the spread. In Indiana, it is illegal to possess live zebra mussels, quagga mussels, or Asiatic clams (312 IAC 9-9-3).

Most management efforts are geared at preventing any further spread of the zebra mussel. Spread is mainly caused by human recreational activities like boating, fishing, and diving. Some simple steps can be taken to help prevent spreading zebra mussels.

- ✓ Remove all plants and animals from your boat, trailer, and accessory equipment before leaving the access area.
- ✓ Drain live wells and bilge water before you leave the access site.
- ✓ Empty bait buckets on land rather than in the water.
- ✓ Wash your boat, tackle, downriggers, and trailer with hot water (above 104°F) when you get home. Flush your motor's cooling system, live wells, bilge and other boat parts that get wet. Let all equipment dry for at least five days before transporting your boat into a new body of water. If planning to move to another body of water sooner, you should disinfect everything that came into contact with water using a 5% bleach solution.
- ✓ Learn to identify the zebra mussel so you can report new sightings. If you find a zebra mussel in a lake that is not currently identified as an invaded lake, preserve the mussel in rubbing alcohol or freeze it, and contact the fisheries biologist in your area for positive identification. Visit the following website to locate your district fisheries biologist:

<http://www.in.gov/dnr/fishwild/fish/fishing/fishbiol.htm>

If you want to find out if there are zebra mussels in a body of water, you can monitor for adults by immersing hard substrate, such as a concrete blocks, in different areas around the lake and check them periodically during summer and fall for attached mussels. You can check any submerged portions of your boat, dock supports, floats, etc. for attached zebra mussels as well.

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Photos courtesy of Sea Grant Great Lakes Network

Updated 12/05

Appendix E
Spotted Lanternfly Quarantine Checklist for Vehicle
Inspection



Spotted Lanternfly Quarantine Checklist for Vehicle Inspection

IMPORTANT: Before traveling from the quarantine area, check for spotted lanternfly egg masses, adults, and nymphs. Make sure your vehicle and all transported items are pest free.
Help stop this pest from spreading!

On May 26, 2018, a new Spotted Lanternfly Order of Quarantine and Treatment was published in the PA Bulletin. The Pennsylvania Department of Agriculture (PDA) established the quarantine order to stop this pest from moving out of the current quarantine zone. PDA is also trying to minimize the movement of SLF within the quarantine zone from areas with higher populations into areas with low to no populations.

The current quarantine zone encompasses these counties: **Berks, Bucks, Carbon, Chester, Dauphin, Delaware, Lancaster, Lebanon, Lehigh, Monroe, Montgomery, Northampton, Philadelphia** and **Schuylkill**. Travel within or out of those counties requires a permit and vehicles must be inspected.

TRAVEL INFORMATION

DATE: _____

DRIVER: _____

VEHICLE: _____

INDICATE COUNTIES TRAVELING IN:

Berks	Lebanon	Other
Bucks	Lehigh	Other
Carbon	Monroe	Other
Chester	Montgomery	Other
Dauphin	Northampton	
Delaware	Philadelphia	
Lancaster	Schuylkill	

INSPECTION

The following is a list of items / places that may harbor SLF. This list is not all-inclusive but should serve as a starting point in actively inspecting for SLF.

- | | | |
|--|-------------------------------------|---|
| ☐ Vehicles (Interior) | ☐ Trailers | ☐ Storage Bins |
| ☐ Vehicles (Exterior) | ☐ Tool Boxes | ☐ Storage Crates |
| ☐ Truck Beds / Caps | ☐ Dollies | |
| ☐ Wheel Wells | ☐ Pallets | |

Please see reverse side for life cycle information.

Depending on the time of year, check for both live insects and / or egg masses. If you find live insects, kill them. If you find egg masses, remove and destroy them.

When inspection is complete, sign and date the reverse side.

SPOTTED LANTERNFLY QUARANTINE CHECKLIST FOR VEHICLE INSPECTION

IMPORTANT: Before traveling from the quarantine area, check for spotted lanternfly egg masses, adults, and nymphs. Make sure your vehicle and all transported items are pest free.
Help stop this pest from spreading!

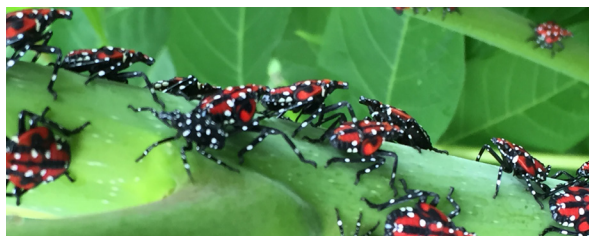
If you find any live insects, kill them. If you find any egg masses, scrape them into a plastic bag, seal the bag, and dispose of it in the garbage.



Adult spotted lanternfly, present in autumn months.



Spotted lanternfly nymphs, present in spring and summer months.



Spotted lanternfly egg mass (outlined in red). Egg masses are present in autumn and winter months, blending in with their surroundings.

By signing this checklist, I am confirming that I have inspected my vehicle and those items I am moving from the Spotted Lanternfly quarantine area, and do not see any egg masses or insects in or on anything I am moving.

Signature: _____ **Date:** _____

For more information, visit the websites of Pennsylvania Department of Agriculture and Penn State Extension.

extension.psu.edu

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