



Animal and Plant Health Inspection Service  
U.S. DEPARTMENT OF AGRICULTURE

# New Pest Response Guidelines

*Cydalima perspectalis*

Box tree moth



Box tree moth adult (Source: Wim Rubers at waarneming.nl, distributed under a CC-BY 3.0 license)

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# Introduction

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Plant Protection and Quarantine (PPQ) develops New Pest Response Guidelines (NPRGs) in preparation for potential pest introductions. This document is based on the best information available at the time of development and may not reflect the latest state of knowledge at the time the pest is detected. In addition, the PPQ response must be tailored to the specific circumstances of each pest introduction event, which cannot be predicted. Therefore, this document provides only general guidelines to be used as a basis for developing a situation-specific response plan at the time a new pest is detected.

Program managers of Federal emergency response or domestic pest control programs must ensure that their programs comply with all Federal Acts and Executive Orders pertaining to the environment, as applicable. Refer to the Environmental Compliance section in [Appendix A](#) for details.

# Pest Overview

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## Key Information

- ◆ Box tree moth, *Cydalima perspectalis*, is native to East Asia and has become a serious invasive pest in Europe, where it continues to spread. In 2018, it was found in Ontario, Canada.
- ◆ The box tree moth feeds primarily on boxwood (*Buxus* spp.).
- ◆ Larvae feed on leaves, and heavy infestations can defoliate host plants. Once the leaves are gone, larvae consume the bark, leading to girdling and plant death.
- ◆ In introduced areas where box tree moth has 2 generations per year, boxwood stands have declined over 95% in 8 years or less.
- ◆ Long-distance spread of the box tree moth is mainly through movement of infested boxwood.
- ◆ The box tree moth can be identified based on morphological characteristics.
- ◆ The most effective way to survey for box tree moths is with bucket traps with pheromone lures.
- ◆ Box tree moth is difficult to eradicate once established but can be effectively controlled with chemical sprays and *Bacillus thuringiensis kurstaki* (Btk).

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## Taxonomy

### Scientific Name

- ◆ *Cydalima perspectalis* (Walker)

### Taxonomic Position

- ◆ Animalia: Arthropoda: Insecta: Lepidoptera: Crambidae

## Synonym(s)

- ◆ *Diaphania perspectalis* (Walker)
- ◆ *Glyphodes perspectalis* (Walker)

## Common Name(s)

- ◆ **Box tree moth**
  - ◆ Box tree pyralid
- 

## Biology and Ecology

The box tree moth is native to temperate and sub-tropical regions in Asia (Mally and Nuss, 2010). It was first reported in Europe in 2007, after which it spread rapidly across Europe into Western Asia and Northern Africa (Agius, 2018; Geci et al., 2020; Haddad et al., 2020; Hizal et al., 2012; Leuthardt and Baur, 2013; Perez and Guillem, 2019). In 2018, it was documented in Canada (Plant et al., 2019).

## Life Cycle

Developmental times for the box tree moth vary with temperature. At 77 °F, the total life cycle (from eggs hatching to adults laying eggs) is between 33 and 44 days (See [Table 2.1](#)). Two to five generations may occur per year, depending on climatic conditions (Wan et al., 2014).

### Eggs

Females lay eggs singly or in clusters of 5 to more than 20 eggs in a gelatinous mass on boxwood leaves (See [Species ID/Diagnostic](#)) (Farahani et al., 2021; Leuthardt and Baur, 2013). Most females deposit more than 42 egg masses in their lifetime (Leuthardt and Baur, 2013). Temperature determines the development time of the eggs (Stan and Mitrea, 2020), but they typically hatch within 4 to 6 days (Farahani et al., 2021).

### Larvae

Box tree moths overwinter as larvae, and all six larval stages are capable of diapausing (Maruyama and Shinkaji, 1991; Nacambo et al., 2014). Overwintering larvae emerge in response to rising temperatures (Poitou et al., 2020). While the timing varies based on local climate, larvae typically begin feeding in March, continuing until they pupate in late April to early May (Farahani et al., 2021; Nacambo et al., 2014; Stan and Mitrea, 2020).

Larvae of the next generation hatch in May or June and immediately begin feeding on boxwood leaves. As they develop, they spin silken webs to hold leaves together and create protected areas to feed (Matošević, 2013; Nacambo et al., 2014). They tend to feed on leaves in the lower portion of host plants, but reside in the upper portion (Kulfan et al., 2020). Larvae remain active through the summer until September or October. Then, in response to shortened daylight hours, larvae will construct a cocoon between two leaves and enter diapause (Xiao et al., 2011).

## Pupae

Pupation occurs on the host leaves in silk cocoons (Korycinska and Eyre, 2011). If the boxwood host is defoliated, pupation may occur away from the host plant using leaves from the surrounding area (Bras, 2021). Pupae will typically first appear in April or May and will be present continuously through the summer and into the fall, depending on the local climate and timing of generations. See [Table 2.1](#) for pupal duration.

## Adults

Adult females use sex pheromones to attract mates (Kim and Park, 2013). They are highly mobile (See [Dispersal](#)) and are typically active at dusk and throughout the night (Székely et al., 2011).

Adults first emerge from the overwintering generation between April and July, depending on climate and temperature. Subsequent generations are active between June and October (Chen et al., 2005; Farahani et al., 2021; Göttig and Herz, 2017; Nacambo et al., 2014). Adults typically live for two weeks after emergence (Bras, 2021). See [Table 2.1](#) for adult lifespan range.

**Table 2-1** Duration of box tree moth life stages and intervals under laboratory conditions at 77 °F (Farahani et al., 2021)

Life stage/interval	Duration (minimum-maximum) in days
Egg	4–6
1 <sup>st</sup> instar larvae	2–5
2 <sup>nd</sup> instar larvae	2–7
3 <sup>rd</sup> instar larvae	2–4
4 <sup>th</sup> instar larvae	2–5
5 <sup>th</sup> instar larvae	3–6
6 <sup>th</sup> instar larvae	5–12
Prepupae	1–2
Total larval period	20–31
Pupae	6–8
Time from Egg to Adult	33–44
Female adult life span	5–27

Life stage/interval	Duration (minimum-maximum) in days
Male adult life span	4–25
Total life span	39–62

## Hosts<sup>1</sup>

The box tree moth completes its development and causes extensive damage mainly on boxwood (*Buxus*) species (John and Schumacher, 2013; Leuthardt and Baur, 2013; Matsiakh et al., 2018; Tuniyev et al., 2016). It has also been shown to complete its development on *Murraya paniculata* (Wang, 2008).

The only other plant species that may support box tree moth development, based on current knowledge, are *Euonymus alatus* and *E. japonicus*, though the available evidence is not conclusive (PPRA, 2021). For *Ilex purpurea* or any other plants on which feeding has been reported in the literature, there is currently no convincing evidence that they support box tree moth development (PPRA, 2021). See [Table 2-2](#) for box tree moth hosts.

**Table 2-2** Box tree moth hosts

Family	Scientific name	Common name	Notes and References	Confidence
Buxaceae	<i>Buxus microphylla</i>	littleleaf boxwood	Maruyama, 1992	Host status certain
Buxaceae	<i>Buxus microphylla</i> var. <i>japonica</i>	Japanese boxwood	Maruyama, 1992	Host status certain
Buxaceae	<i>Buxus sempervirens</i>	common boxwood	Hizal, 2012	Host status certain
Buxaceae	<i>Buxus sinica</i> var. <i>insularis</i> (= <i>Buxus microphylla</i> var. <i>insularis</i> )	Korean boxwood	Maruyama, 1992	Host status certain
Rutaceae	<i>Murraya paniculata</i>	mock orange	Wang, 2008	Host status certain
Celastraceae	<i>Euonymus alatus</i>	burningbush	Larvae are documented feeding on this plant in the field (Chen and Chen, 2017; Tu et al., 2017; Zhang and Wang, 2012), but laboratory results are mixed. Two laboratory tests showed minimal feeding and no development (Brua, 2013; Wiesner et al., 2021). One report stated adults were reared from larvae that were fed this plant, but no details were provided (Brua, 2013).	Host status uncertain

<sup>1</sup> Hosts are plants that support box tree moth development.

Family	Scientific name	Common name	Notes and References	Confidence
Celastraceae	<i>Euonymus japonicus</i>	Japanese spindle tree	<p>Most reports refer to this plant using a Chinese common name that is also used for <i>Buxus megistophylla</i> (Chen, 2008; Liu et al., 2009; Ma and Yang, 2010; Qi, 2003; Sun et al., 2009; Wang et al., 2013).</p> <p>Larvae are documented feeding on this plant in the field (Chen, 2008; Gu and Yang, 2006; Liu et al., 2009; Ma and Yang, 2010; Qi, 2003; Sun et al., 2009; Wang et al., 2013). Laboratory studies, however, have shown that this plant does not support complete development (Brua, 2013; Karpun et al., 2015; Matošević et al., 2017; van der Straten and Muus, 2010).</p>	Host status uncertain

## Dispersal

The rate of spread for the box tree moth has varied since its introduction to Europe, with some cases peaking at 96 miles per year (Roques et al., 2016).

### Human-Assisted Spread

Long distance movement of the box tree moth across Europe occurred through the movement of infested boxwood plantings (Bras et al., 2019). Multiple detections have been made at nurseries where boxwoods are commonly grown and shipped over long distances (Bella, 2013).

### Natural Dispersal

Box tree moths are highly mobile and are reported to be good fliers (Bella, 2013). Natural spread of this moth in Europe is about 3-6 miles per year (John and Schumacher, 2013; van der Straten and Muus, 2010). One analysis from Europe concluded that natural dispersal from continental Europe to the United Kingdom was possible, suggesting sustained adult flights of over 20 miles (Korycinska and Eyre, 2011).

## Species ID/Diagnostic

### Morphological

#### Adults

Bodies are white, with a brown head and abdomen (Fig. 3-1) (Matošević, 2013). Wings are white and slightly iridescent, with an irregular thick brown border spanning 1.6–1.8 inches (Gutue et al., 2014; Korycinska and Eyre, 2011). Some adults have completely brown wings with a small white streak on each forewing (Fig. 3-2) (Albert, 2009; Korycinska and Eyre, 2011). Males and females show both colorations (Bras, 2021).



**Figure 3-1** Adult male box tree moth (Source: Szabolcs Sáfíán, Bugwood.org)



**Figure 3-2** Adult male brown box tree moth (Source: Szabolcs Sáfián, Bugwood.org)

## Eggs

Eggs are pale yellow, averaging 0.04 inches and laid in flat clusters (Fig. 3-3) (Hizal et al., 2012; Korycinska and Eyre, 2011). As they mature, a black spot appears marking the larval head (Korycinska and Eyre, 2011).



**Figure 3-3** Eggs of box tree moth on a boxwood leaf (Source: W. Schön, [www.schmetterling-raupe.de](http://www.schmetterling-raupe.de))

## Larvae

Newly hatched larvae have black heads and are green to yellow (Fig. 3-4). As they age, dark brown stripes develop on the body. 6<sup>th</sup> instar larvae are about 1.6 inches long and have thin white and thick black stripes and black dots outlined in white along the length of the body (Fig. 3-5) (Korycinska and Eyre, 2011).



**Figure 3-4** Newly hatched box tree moth larvae (Source: Colette Walter, <http://www.schmetterling-raupe.de/art/perspectalis.htm>)



**Figure 3-5** Box tree moth larvae (Source: Böhringer Friedrich, at [rufre@lenz-nenning.at](mailto:rufre@lenz-nenning.at), distributed under a CC-BY 3.0 license)

## Pupae

Pupae develop inside a silk cocoon and are 0.6–0.8 inches long. They are initially green, with black stripes on the back, and turn brown as they mature (Fig. 3-6) (Korycinska and Eyre, 2011).



**Figure 3-6** (A and B) Box tree moth pupae surrounded by white silk and host foliage; (C and D) pupae exposed after removal of white silk and host foliage (Sources: Korycinska and Eyre, 2011 and <http://www.schmetterling-raupe.de/art/perspectalis.htm>)

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## Signs and Symptoms

Signs of damage may not appear at the beginning of an infestation because young larvae hide among twigs and leaves (Hrnčić and Radonjić, 2017). Larvae skeletonize the leaves and feed on the bark, causing defoliation and dryness, leading to the host's death (Figs. 3-7 and 3-8) (Hrnčić and Radonjić, 2017; Nacambo et al., 2014). Signs of feeding include green-black frass and webbing (Fig. 3-9) (Gutue et al., 2014).



**Figure 3-7** Mature box tree moth larva feeding on leaves of common boxwood (Source: Ferenc Lakatos, University of West-Hungary, Bugwood.org)



**Figure 3-8** Box tree moth damage to common boxwood (Source: Ferenc Lakatos, University of West-Hungary, Bugwood.org)



**Figure 3-9** Common boxwood defoliation and webbing due to box tree moth feeding (Source: Colette Walter, [http://www.lepiforum.de/webbbs/images/forum\\_2/pic13983.jpg](http://www.lepiforum.de/webbbs/images/forum_2/pic13983.jpg))

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## Similar Species

The melonworm, *Diaphania hyalinata*, is found in the United States in south Florida and possibly south Texas and feeds on the leaves of cucurbits (Fig. 3-10) (Capinera, 2020). The adult moth is similar in appearance to the box tree moth.



**Figure 3-10** Adult melonworm (Source: Jack Foreman, distributed under a CC-BY 3.0 license)

Boxwood dieback caused by the fungal pathogen, *Colletotrichum theobromicola*, causes dieback of twigs with light tan leaves that remain attached to the branches (Singh and Doyle, 2017). The infection also causes a black discoloration under the bark of twigs (Singh and Doyle, 2017). This disease of boxwoods has been found in Louisiana, North Carolina, South Carolina and Virginia (Singh et al., 2015).



**Figure 3-11** Boxwood dieback of twigs (A) with dying leaves, and (B) black discoloration of stem under bark (Source: Raj Singh, LSU AgCenter)

# Delimitation Survey

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## Delimitation Area

Delimitation surveys determine the extent of the infested area after a pest has been found. The size and shape of the delimitation area may differ from one location to another due to differences in host plant density and distribution, topography, and agency resources at the time of pest introduction.

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## Timing of Surveys

Start the delimitation surveys immediately after detecting box tree moth. Depending on the climatic conditions where the moth is detected, two to five generations may occur per year. If no additional detections are made, surveys should continue for at least one year in areas with three or more generations and for two years in areas with two generations.

Survey continuously when temperatures are greater than 51°F and adults are flying, typically between May and October. If moths are detected in the fall in areas where the pest is likely to overwinter, surveys should resume the following spring, based on degree day models for adult emergence.

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## Type of Survey

The most suitable method for detecting and delimiting box tree moth is through the use of pheromone traps. The survey design focuses on trapping in the core area around the initial detection and along the perimeter of the survey area.

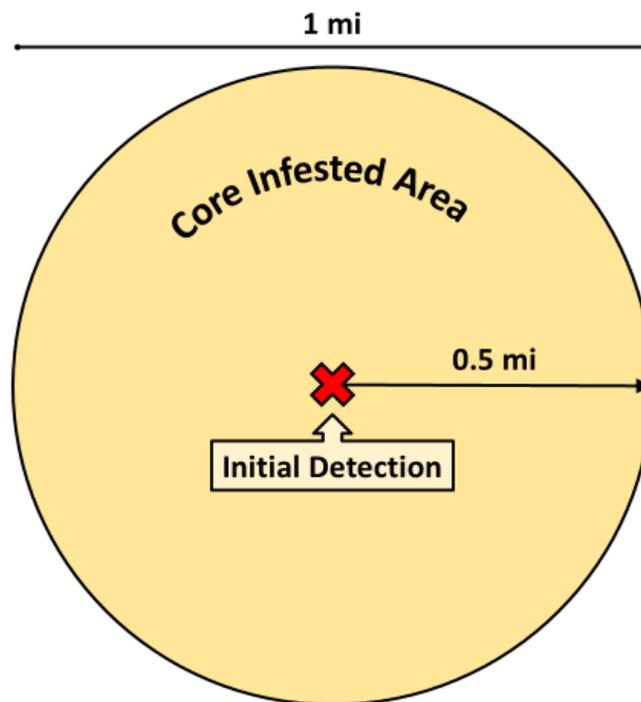
Visual inspections for box tree moths and damage to host plants (see [Signs and Symptoms](#) for descriptions) can be useful, but some infested plants may not have visible insects or symptoms, depending on the time and severity of infestation.

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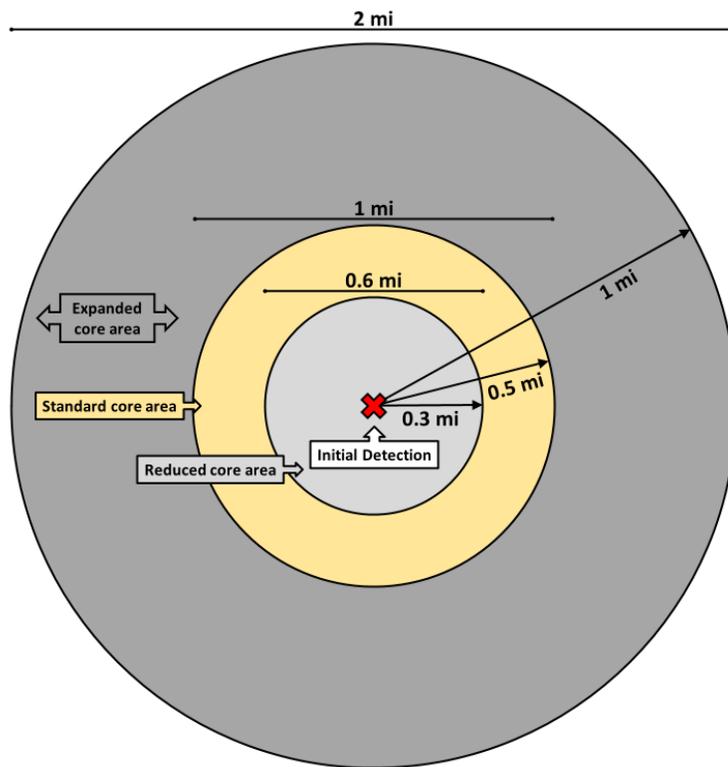
## Delimitation Survey Design

To delimit box tree moth using a core and transect design:

1. Identify the core infested area and the extended survey transects.
  - a. Using a map or mapping software, draw a circle that extends out from the initial detection (Fig. 4-1).
    - i. Standard surveys should have a core infested area extending 0.5 miles from the initial detection.
    - ii. Under certain scenarios, a smaller (0.3 mile radius) or larger (1 mi radius) core infested area will be adequate (Fig. 4-2).
      1. Use a smaller core if resources are limited or traps are not available and a larger core if enough resources are available.
      2. Use a smaller core if the detection is made where moths are unlikely to disperse (e.g., in a nursery where there are plentiful host plants).
      3. Use a larger core if the pest population is well-established near the initial detection and is more likely to have dispersed long distances.

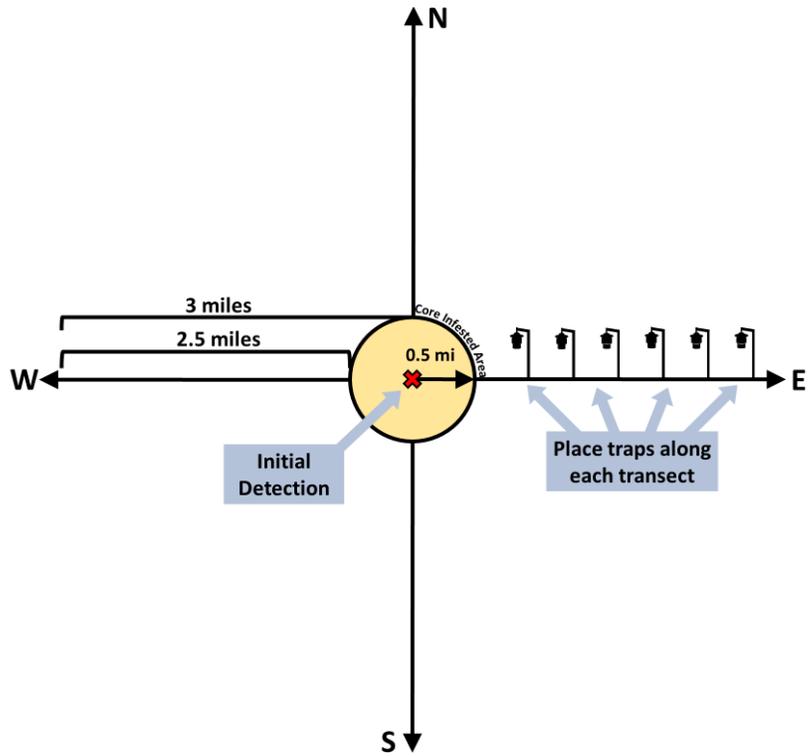


**Figure 4-1** Standard core infested area after an initial detection

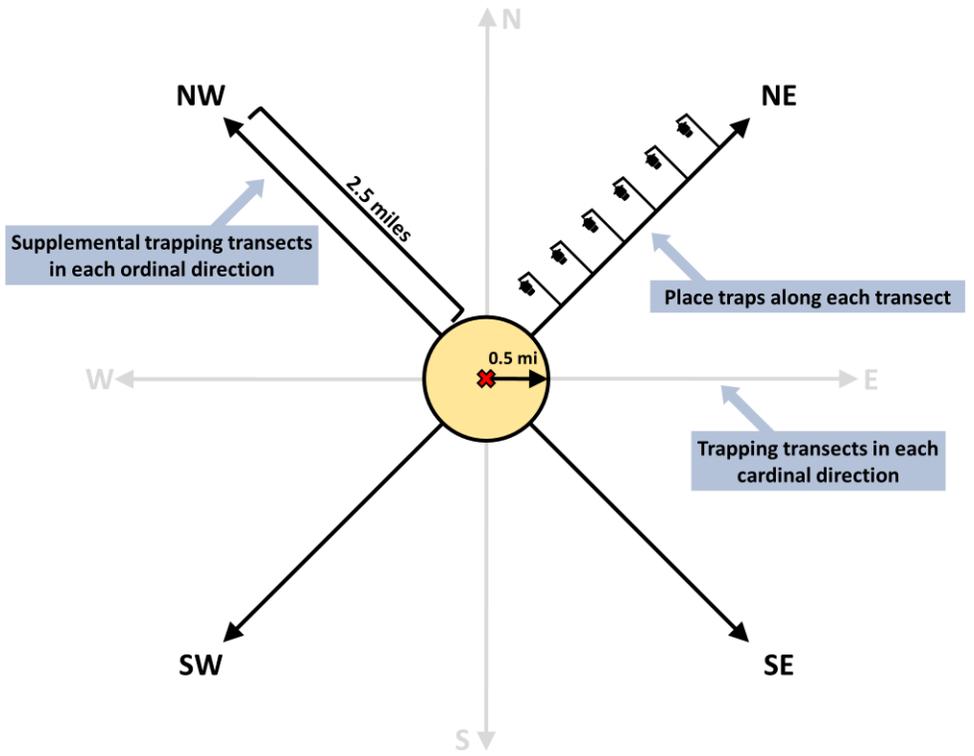


**Figure 4-2** Alternate core infested area sizes

- b. Using a map or mapping software, draw four transect lines.
  - i. If moths are detected at a retailer or nursery, surveyors may use either the retailer’s service area if that information is available to determine the length and direction of surveying transects.
  - ii. If there isn’t any information to suggest a direction for transects, place them in each cardinal direction (Fig. 4-3).
  - iii. If more traps are available and if there is no information available regarding the extent or direction of spread, consider drawing four additional transect lines in each of the ordinal directions (NE, SE, NW, SW) (Fig. 4-4).
  - iv. Standard transect length should be 3 miles from the initial detection.
    1. A standard core area with radius of 0.5 miles will have 2.5-mile transects.
    2. A smaller core area (0.3-mile radius) will have 2.7-mile transects and a larger core (1-mi radius) will have 2-mile transects.

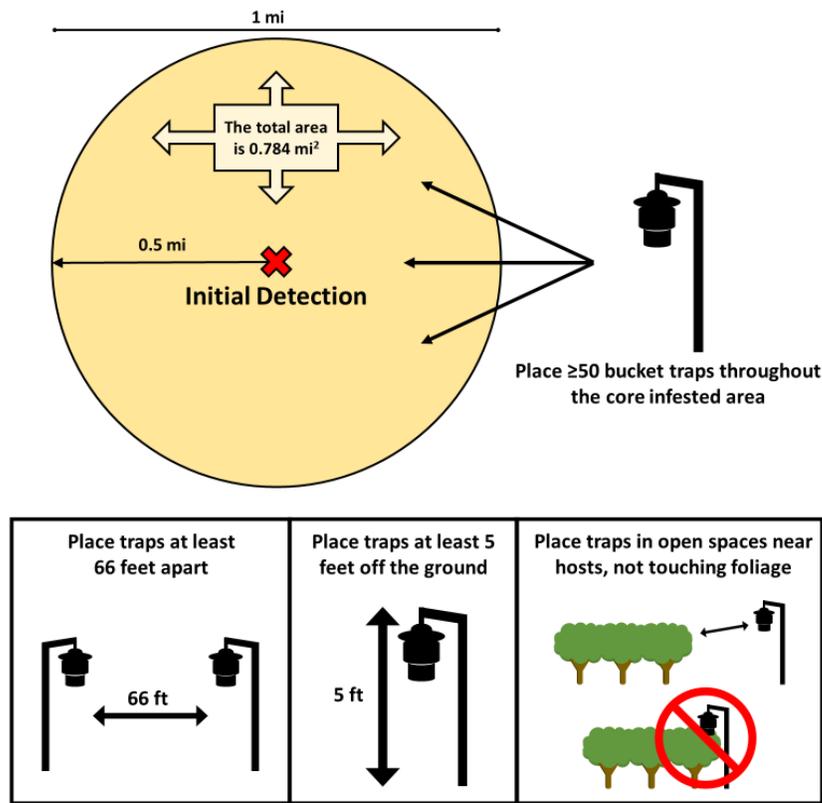


**Figure 4-3** Trapping transects extending out from a core infested area in the cardinal directions



**Figure 4-4** Additional trapping transects extending out from a core infested area in the ordinal directions

2. Survey the core infested area.
  - a. Place traps throughout the core infested area at a density of 64 traps per square mile<sup>2</sup>.
  - b. A core infested area with a radius of 0.5 miles covers 0.784 square miles, which means you will need at least 50 traps to survey this area (Fig. 4-5). A 0.3-mile radius will require at least 18 traps and a 1-mile radius will require at least 201 traps.
  - c. Space traps regularly throughout the area, at least 66 feet apart, and near hosts when possible. See the [Sampling Instructions](#) for details.
  - d. Traps should be checked at least every two weeks and should be kept in place according to the guidelines in [Timing of Surveys](#).



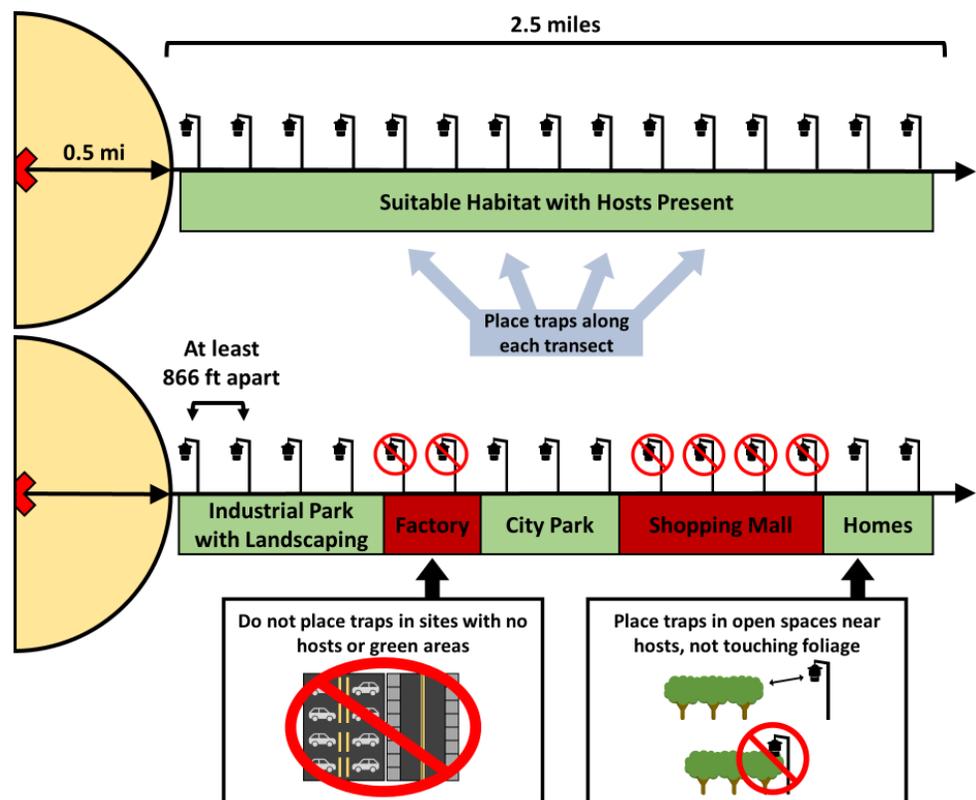
**Figure 4-5** Placing traps in a core infested area with a 0.5 mi radius. Trapping density is 64 traps per square mile. Traps needed will change with a smaller or larger core.

3. Survey along the transect lines (Fig. 4-6).
  - a. Each 2.5 mi transect requires up to 15 traps, based on the trap-to-trap distance when using 36 traps per square mile. Consult the survey design tool (included separately) to adjust the trap placement to fit the

<sup>2</sup> Consult the survey design tool (included separately) to see the probabilities of detection for different trap densities.

needs of the survey.

- b. For a 2.5 mi transect with 36 traps per linear mile, place traps at least 866 feet apart. Consult the survey design tool (included separately) to adjust the trap spacing with the change in trap density.
- c. Only place traps in areas with available hosts.
- d. Barren areas or non-permeable surfaces should be excluded. Do not place traps in these areas.
- e. If areas with continuous hosts and suitable habitats are available along the entire transect, surveyors will deploy 15 traps spaced evenly.
- f. Up to 60 traps are required to fully cover transects in each cardinal direction (Fig. 4-3). Up to 120 traps are required to fully cover transects placed in both the cardinal and ordinal directions (Fig. 4-4).
- g. Follow the [Sampling Instructions](#) for details on trap placement.
- h. Traps should be checked at least every two weeks and should be kept in place according to the guidelines in [Timing of Surveys](#).



**Figure 4-6** Placing traps along a trapping transect. Place traps in habitat with hosts and avoid placing traps in unsuitable areas. If suitable areas are continuous along the 2.5 mile transect, 15 traps will be used.

4. Employ an [Outreach Program](#) to raise awareness and detect box tree moth over larger areas where surveys are not feasible.

## Survey Adjustments and Expansion

### Adjustments

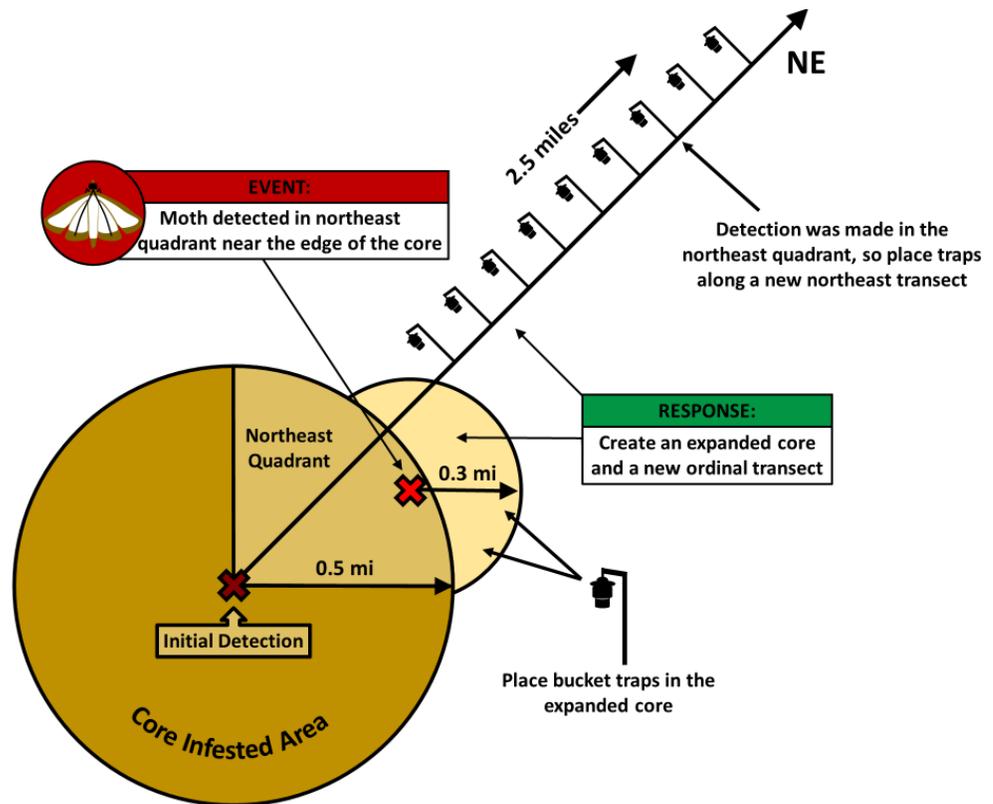
As data become available after traps in the field are serviced, surveyors may need to adjust their trapping protocols. If there are no detections in the transect, consider changing the location of the transects to cover more area outside the core. If there are no detections in the core, continue trapping in the core for about three months to capture potential next generation adults. After confirming that there are no new populations in the core, the traps may be moved to areas outside the core. If moths are found, surveyors can adjust the trapping density in the core to focus on specific locations or add/extend trapping transects to better delimit pests near additional detections outside the core.

To more effectively place additional traps, consider investigating whether moths found in the core or along trapping transects originated from nearby areas. Surveyors may look for infested hosts around the trap or at high-risk sites such as retailers or nurseries in the vicinity of the trap (see [Signs and Symptoms](#) for images of infestations). If any infested hosts are found, surveyors can adjust trapping locations or density based on the source of infestation or direction of spread, as determined by trace-back and trace-forward surveys.

### Survey Expansion in the Core Area

If any new detections are made within 0.3 miles of initial detection, do not expand the survey. Continue trapping in the area, following the guidelines from [Timing of Surveys](#). Detections close to the initial detection will inform control efforts by determining pest density and dispersal direction.

If any detections are made between 0.3 miles and 0.5 miles of the initial detection, expand the core from the point of additional detection in a circle with a radius of 0.3 miles ([Fig 4-7](#)). Place traps throughout the expanded core infested area at a density of 64 traps per square mile. In addition, add an ordinal trapping transect (NE, NW, SE, SW) in the quadrant where the detection was made, if ordinal transects have not already been placed.

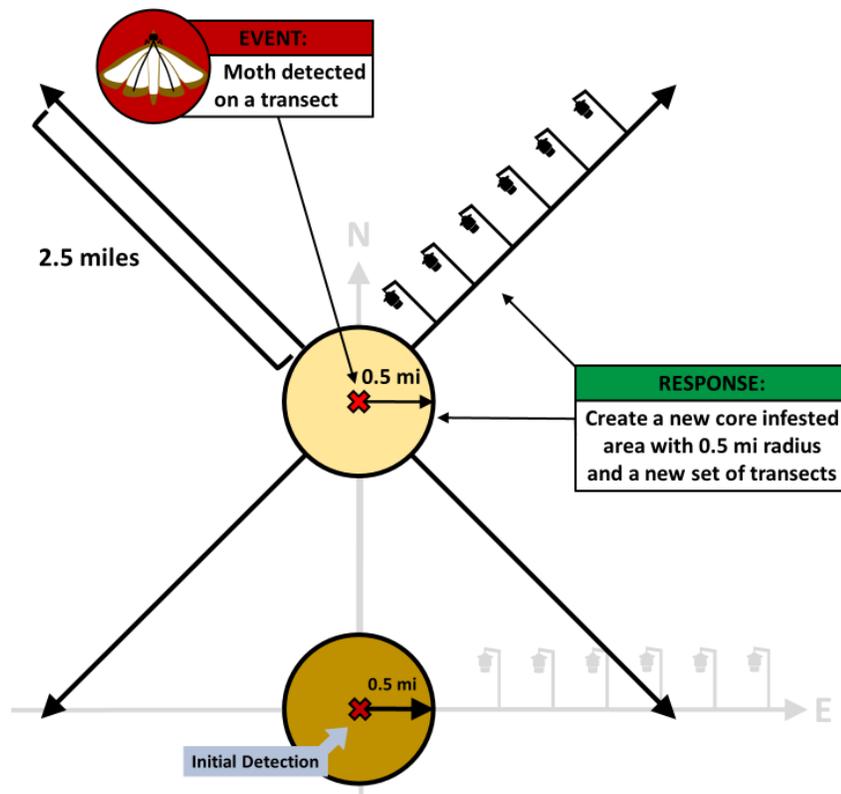


**Figure 4-7** Survey expansion after detecting moths in the core infested area. Surveyors should add an ordinal transect if one has not already been included in the survey.

### Along the Trapping Transects

Additional detections along trapping transects may indicate a new focal point of the infestation and require survey expansion. After any detection from a trapping transect, use mapping software to create an additional core infested area (extending 0.5 miles from the new detection) and a new set of trapping transects extending 3 miles from the detection).

Surveyors may need to adjust the size of the core, the length of the trapping transects, and the direction of the trapping transects based on the available moth distribution data. Do not place additional traps in previously surveyed areas. If the detection is relatively close to the initial detection, placing transects at different angles will help cover unsurveyed areas (Fig. 4-8).



**Figure 4-8** Survey expansion after detecting moths in the trapping transects. Expanded trapping transects are placed at 45 degree angles to the original transects to better cover the area.

## Outreach Program for Box Tree Moth

Developing an outreach campaign as a supplement for traditional surveys will more effectively detect box tree moth over a large area. This outreach should be organized and carried out by state extension services and operate side by side with traditional detection surveys by PPQ staff. This campaign should target surveyed and at-risk areas near the initial detection. It should encourage nurseries and landscape managers in the region to proactively inspect hosts, place sentinel traps around hosts, and report infestations (see [Signs and Symptoms](#)) to the proper authorities. Photographs of box tree moths on plants should be submitted to local extension agents or PPQ staff to help them identify new detections and areas for further investigation.

# Eradication and Control Options

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## Overview

This information can be used by PPQ decision-makers after a detection to assess the suitability of potential actions to eradicate, contain, or suppress box tree moth. Although eradication should always be prioritized, its success will depend on the pest population size and distribution. Use the chemical and Bt-based options listed below to attempt eradication of the pest.

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## Eradication Options

There is no evidence of successful eradication of the box tree moth where it has been introduced (CABI/EPPO, 2018). Eradication of established populations in Europe, including in Germany and Switzerland, is no longer considered feasible (Albert, 2009; van der Straten and Muus, 2010). However, eradication is possible if the pest is contained soon after introduction in smaller patches of cultivated or highly managed boxwoods. In these cases, chemical sprays should be used to kill larvae on infested hosts. In areas where the moth has become widely established, use the control options listed below to contain or suppress the box tree moth.

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## Control Options

### Host Removal

If heavily infested boxwoods are visually detected, remove and destroy the infested shrubs and any nearby hosts to prevent further rapid spread of the moth.

- ◆ Hosts can be destroyed by burial, chipping, or burning. In case of burning, check local ordinances for guidelines and required documentation.
- ◆ Treat uninfested and unremoved hosts prophylactically with a pyrethroid or organophosphate (see [Chemical Control](#), below).

Be sure to contain affected plants to safeguard the material while it is being

moved to another site for destruction, especially when chemical control is not feasible. Placing plant material (e.g. potted or removed plants) in large plastic bags may prevent insect escapes. Placing bagged material in the sun will increase temperatures that may be lethal to the insect.

## Chemical Control

Chemical controls may require high spray volume for effective control of box tree moth since the caterpillars feed on the undersides of leaves and create webbing that may protect them from adequate exposure. In Asia, pyrethroids (deltamethrin, cypermethrin), organophosphates (chlorfluazuron), spinosyns (spinosad), and phenylpyrazoles (fipronil) are used against box tree moth (Wan et al., 2014). Spinosad and fipronil are recommended treatments in parts of China (Wan et al., 2014) and are most effective against early larval stages (Kenis, 2016). In infested areas of Europe, chemical treatments have been the primary means to protect *Buxus* plantings (Kenis et al., 2013). Table 5-1 displays the most effective active ingredients tested against box tree moth larvae. Insecticide options are not available to use on pupae.

**Table 5-1** Insecticides available in the United States for use against box tree moth

IRAC Insecticide Class (Mode of Action) <sup>1</sup>	Active Ingredient	Efficacy	Registered in the U.S.*	References for use
Diamides (28)	Chlorantraniliprole	99%	yes	Somsai et al., 2019
Ecdysone Receptor Antagonists (18)	Methoxyfenozide	99%	yes	Somsai et al., 2019
Glutamate gated chloride channel blocker (GluCl) (6)	Abamectin	100%	yes**	Somsai et al., 2019
Glutamate gated chloride channel blocker (GluCl) (6)	Emamectin benzoate	>95%	yes	Qian et al., 2018
Neonicotinoids (4A)	Thiacloprid	96%	yes***	Stan and Mitrea, 2019
Organophosphate (1B)	Trichlorfon	>95%	yes**	Qian et al., 2018
Organophosphate (1B)	Dimethoate	100%	yes	Raspudić et al., 2018
Organophosphate (1B)	Chlorpyrifos	93%	yes	Somsai et al., 2019
Pyrethroids (3A)	λ-cyhalothrin	98%	yes	Stan and Mitrea, 2019
Pyrethroids (3A)	β-cyfluthrin	>95%	yes	Qian et al., 2018
Pyrethroids (3A)	Cypermethrin	98%	yes	Stan and Mitrea, 2019
Pyrethroids (3A)	τ-Fluvalinate	98%	yes**	Stan and Mitrea, 2019
Spinosyns (5)	Spinosad	99%	yes	Somsai et al., 2019

<sup>1</sup> MOA: Insecticide acts on nerve & muscle (1B, 3A, 4A, 5, 6, 28), or acts as an insect growth regulator (18). For full definitions for each mode of action see <https://irac-online.org/modes-of-action>.

\*Registration information collected from CDMS, 2021. No active ingredients are labeled specifically for box tree moth.

\*\*Registered in the United States, but not typically labeled for lepidopteran pests.

\*\*\*Registered in the United States, with no commercial products available.

## Biological Controls

### Egg

*Chelonus tabonus*, *Tyndarichus* spp. and *Trichogramma* spp. are reported to parasitize box tree moth eggs (Göttig and Herz, 2016; Wan et al., 2014).

### Larva

Bioinsecticides based on *Bacillus thuringiensis* serotype *kurstaki* (Btk) have been effective against box tree moth larvae in Europe (Guérin, 2018) and are now the recommended control method in that region (Göttig and Herz, 2018; Lefort et al., 2014).

*Casinarina* spp., *Compsilura concinnata*, *Dolichogenidea stantoni*, *Exorista* sp., *Protapanteles mygdonia* and *Pseudoperichaeta nigrolineata* are larval parasitoids of box tree moth (Belokobylskij and Gninenko, 2016; Farahani et al., 2018; Shi and Hu, 2007; Wan et al., 2014). In laboratory studies, entomopathogenic nematodes *Steinernema carpocapsae* and *Heterorhabditis bacteriophora* caused high mortality of box tree moth larvae.

### Pupa

*Brachymeria lasus* and *Apechthis compunctator* are pupal parasitoids of box tree moth (Wan et al., 2014).

## Cultural Controls

Removing larvae manually or via water-spraying has been recommended for control on ornamental box trees, but not for forests or other large patches of unmanaged hosts (Alkan Akıncı and Kurdoğlu, 2019; Wan et al., 2014). Manual removal is labor intensive, and water-spraying is difficult and may damage the tree (Kenis, 2016). Recommendations for parks and gardens in China include visual inspection and removal of eggs every two to three days during the oviposition season (Shi and Hu, 2007).

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# Environmental Compliance

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## Introduction

Use Appendix A as a guide to environmental regulations pertinent to the box tree moth.

## Overview

Program managers of Federal emergency response or domestic pest control programs must ensure that their programs comply with all Federal Acts and Executive Orders pertaining to the environment, as applicable. Two primary Federal Acts, the National Environmental Policy Act (NEPA) and the Endangered Species Act (ESA), often require the development of significant documentation before program actions may commence. Environmental and Risk Analysis Services (ERAS), a unit of APHIS' Policy and Program Development Staff (PPD), is available to provide guidance and advice to program managers and prepare drafts of applicable environmental documentation. In preparing draft NEPA documentation ERAS may also perform and incorporate assessments that pertain to other Acts and Executive Orders, described below, as part of the NEPA process. The Environmental Compliance Team (ECT), a part of PPQ's Plant Health Programs (PHP), assists ERAS in development of documents and implements any environmental monitoring. Program leadership is strongly advised to consult with ERAS and/or ECT early in the development of a program in order to conduct a preliminary review of applicable environmental statutes and to ensure timely compliance.

Environmental monitoring of APHIS pest control activities may be required as part of compliance with environmental statutes, as requested by program managers, or as suggested to address concerns with controversial activities. Monitoring may be conducted with regards to worker exposure, pesticide quality assurance and control, off-site chemical deposition, or program efficacy. Different tools and techniques are used depending on the monitoring goals and control techniques used in the program. Staff from ECT will work with the program manager to develop an environmental monitoring plan, conduct training to implement the plan, provide day-to-day guidance on monitoring, and provide an

interpretive report of monitoring activities.

The following is list of pertinent laws and Executive Orders:

**National Environmental Policy Act (NEPA)** – NEPA requires all Federal agencies to examine whether their actions may significantly affect the quality of the human environment. The purpose of NEPA is to inform the decision-maker prior to taking action and to inform the public of the decision. Actions that are excluded from this examination, actions that normally require an Environmental Assessment, and actions that normally require Environmental Impact Statements are codified in APHIS' NEPA Implementing Procedures located in 7 CFR 372.5.

The three types of NEPA documentation are:

### **1. Categorical Exclusion**

Categorical exclusions are classes of actions that do not have a significant effect on the quality of the human environment and for which neither an environmental assessment (EA) nor an environmental impact statement (EIS) is required. Generally, the means through which adverse environmental impacts may be avoided or minimized have actually been built into the actions themselves (see 7 CFR 372.5(c)).

### **2. Environmental Assessment (EA)**

An EA is a public document that succinctly presents information and analysis for the decision-maker of the proposed action. An EA can lead to the preparation of an environmental impact statement (EIS), a finding of no significant impact (FONSI), or the abandonment of a proposed action.

### **3. Environmental Impact Statement (EIS)**

In the event that a major Federal action may significantly affect the quality of the human environment (adverse or beneficial), or, the proposed action may result in public controversy, an EIS is prepared.

**Endangered Species Act (ESA)** – This statute requires that programs consider their potential effects on federally protected species. The ESA requires programs to identify protected species and their habitat in or near program areas and documentation of how adverse effects to these species will be avoided. The documentation may require review and approval by the U.S. Fish and Wildlife Service and the National Marine Fisheries Service before program activities can begin. Knowingly violating this law can lead to criminal charges against individual staff members and program managers.

**Migratory Bird Treaty Act** – This statute requires that programs avoid harm to

over 800 endemic bird species, eggs, and their nests. In some cases, permits may be available to capture birds, which require coordination with the U.S. Fish and Wildlife Service.

**Clean Water Act** – This statute requires various permits for work in wetlands and for potential discharges of program chemicals into water. This may require coordination with the Environmental Protection Agency, individual states, and the Army Corps of Engineers. Such permits would be required even if the pesticide label allows for direct application to water.

**Tribal Consultation** – This Executive Order requires formal government to government communication and interaction if a program might have substantial direct effects on any federally-recognized Indian Nation. This process is often incorrectly included as part of the NEPA process, but must be completed prior to general public involvement under NEPA. Staff should be cognizant of the conflict that could arise when proposed federal actions intersect with tribal sovereignty. Tribal consultation is designed to identify and avoid such potential conflict.

**National Historic Preservation Act** – This statute requires programs to consider potential impacts on historic properties (such as buildings and archaeological sites) and requires coordination with local State Historic Preservation Offices. Documentation under this act involves inventorying the project area for historic properties and determining what effects, if any, the project may have on historic properties. This process may require public involvement and comment prior to the start of program activities.

**Coastal Zone Management Act** – This statute requires coordination with states where programs may impact Coastal Zone Management Plans. Federal activities that may affect coastal resources are evaluated through a process called “federal consistency”. This process allows the public, local governments, Tribes, and state agencies an opportunity to review the federal action. The federal consistency process is administered individually by states with Coastal Zone Management Plans.

**Environmental Justice** – This Executive Order requires consideration of program impacts on minority and economically disadvantaged populations. Compliance is usually achieved within the NEPA documentation for a project. Programs are required to given consider if the actions might disproportionately impact minority or economically disadvantaged populations, and if so, how such impact will be avoided.

**Protection of Children** – This Executive Order requires federal agencies to identify, assess, and address environmental health risks and safety risks that may disproportionately affect children. If such a risk is identified, then measures must

be described and implemented to minimize such risks.

# Sampling

## Insect Collection: Trapping

### Pheromone traps

1. Trap set-up
  - ◆ Use bucket traps (Unitraps) and follow the [Bucket Trap Protocol](#).
  - ◆ Use the pheromone combination 5:1 (Z)-11-hexadecenal:(E)-11-hexadecenal.
  - ◆ Load 0.000002 ounces of pheromone onto laminate pheromone dispensers (photo-setting resin on polypropylene film). Laminate dispensers are more effective than rubber septa dispensers.
  - ◆ Change or re-charge dispensers with new pheromones every 1 to 2 weeks.
2. Trap placement and maintenance
  - ◆ Traps should be placed at least 66 feet apart.
  - ◆ Position traps roughly five feet above the soil surface.
  - ◆ Place traps near hosts whenever possible, but away from foliage (See [Bucket Trap Protocol](#)).
  - ◆ Service the trap every 2 weeks (Kim and Park, 2013).

### Other available trapping techniques

- ◆ Light traps (Nacambo et al., 2014).
- ◆ Feeding attractant traps (phenylacetaldehyde-based and iso-amyl alcohol-based lures) (Nagy et al., 2017).

### Trapping in highly developed areas

Surveyors will need to consider the potential challenges when adjusting this survey to fit highly developed urban or residential landscapes with few suitable or accessible trapping sites. Optimizing the likelihood of capture by placing traps around possible hosts or in green areas near high-risk introduction points should remain the priority. High-risk areas will often include airports, warehouses, nurseries, or other sites that are likely to move high volumes of host plants and other products that can carry pests. Surveying around these high-risk areas will

require on the ground assessments for access and site suitability as well as permissions from property owners to place and maintain traps. With permissions in place, trapping along roadsides, in parking lots, and in parks/green areas are likely to be essential for proper coverage in an urban landscape.

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## **Sample Submission**

Contact your State Plant Health Director for sample submission guidance.

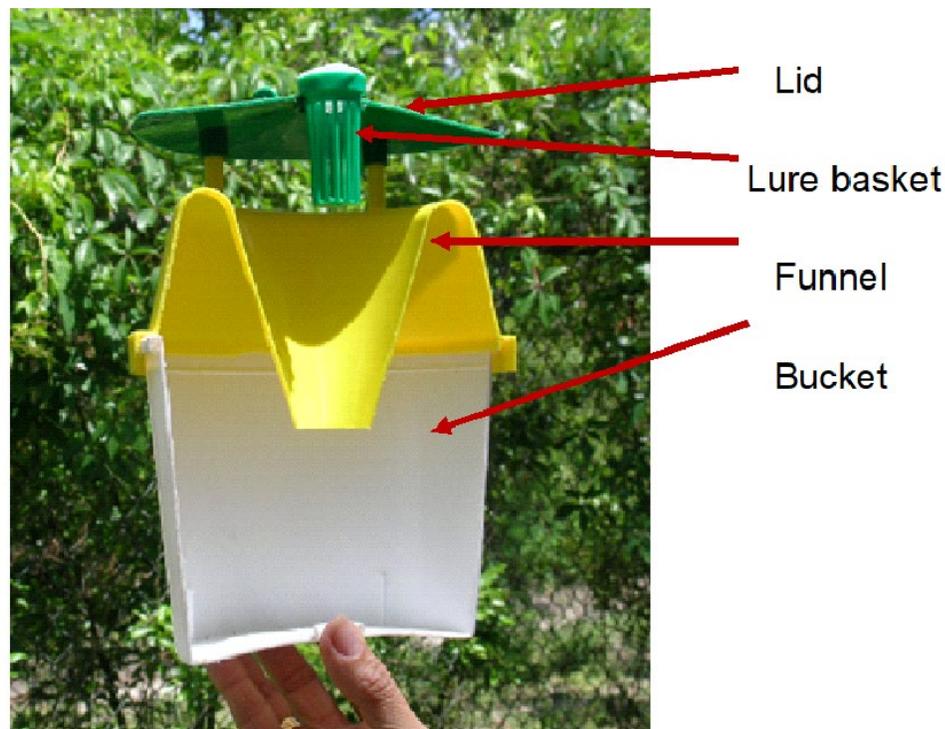
# Plastic Bucket Trap Protocol



## Plastic Bucket Trap Protocol



The plastic bucket trap is a long-lasting insect trap used in conjunction with a lure to monitor or detect various species of moths. The plastic bucket trap is the preferred trap for some moth species since it can catch large numbers of moths without damaging some of their identifying characters. The trap has four parts: 1) lid, 2) lure basket with cap, 3) funnel, and 4) bucket. The trap is available in various color combinations. For PPQ programs, the trap consists of a green lid, yellow funnel, and white bucket. **Figure 1** is a photograph of a trap cut in half.

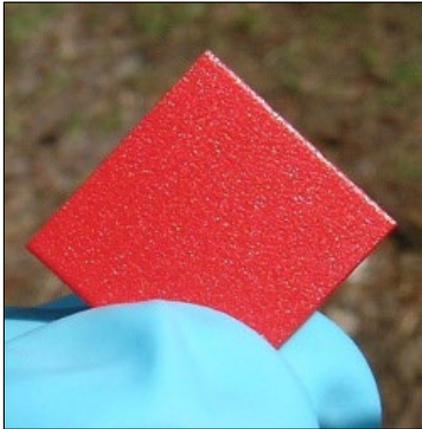


**Figure 1** Plastic bucket trap cut in half to show its interior

**Follow the steps below to prepare the bucket traps for use in the field.**

### **1. Pheromone**

The synthetic pheromone is embedded in a laminate (a small rubberized square as in **Figure 2**) or a septum (similar to a pencil eraser as in **Figure 3**) dispenser. Use gloves when handling lures and unwrap the lure from its packaging. When not in use, the lures should be stored in a freezer not used for food or drinks. Refer to the pheromone MSDS documents for storage and safety information.



**Figure 2** Laminate lure



**Figure 3** Septum lure

If you are using a laminate lure, you may attach the lure to a small paper clip and fold the clip so that the lure does not fall out of the basket (**Figures 4 and 5**).



**Figure 4** Laminate lure in paper clip



**Figure 5** Laminate lure in paper clip inside lure basket

If you are using a septum lure, place the septum inside the lure basket (**Figure 6**). Cover the basket with the lid and insert the basket through the circular opening on the center of the lid (**Figure 7**). If the cap no longer snaps snugly into the trap lid opening, secure it with a piece of tape.



**Figure 6** Septum lure inside lure basket

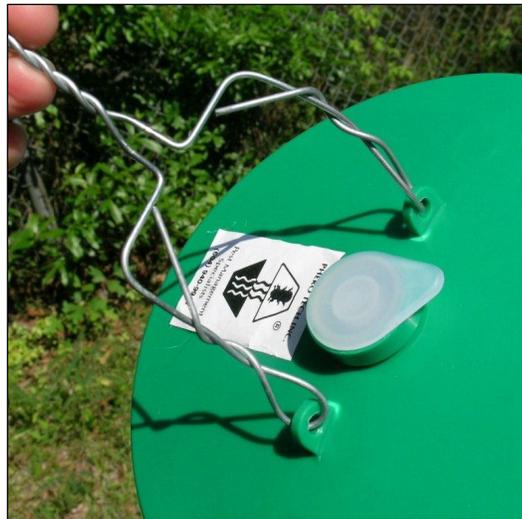
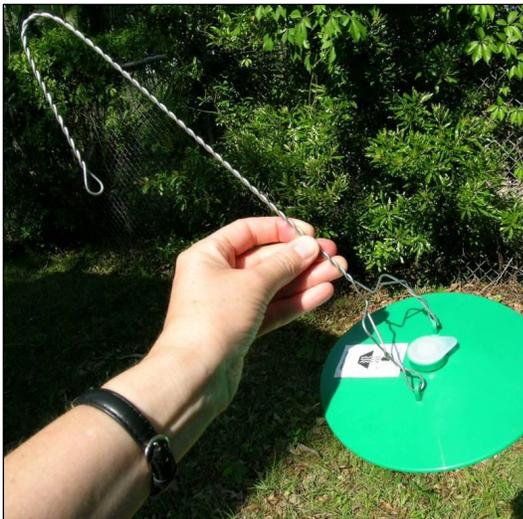


**Figure 7** Lure basket with cap inserted through center of lid

**Note:** To avoid cross-contamination from lure residue, do not reuse lure baskets for different species. Label the lure basket with the name of the moth species.

## 2. Handle

Attach a wire handle to the lid through its two loops, as shown in **Figures 8 and 9**. A wire handle is usually included with each trap. If a handle is not included or needs to be replaced, make one with a 12-inch long wire. You can also use string, but it does not last as long as the wire.



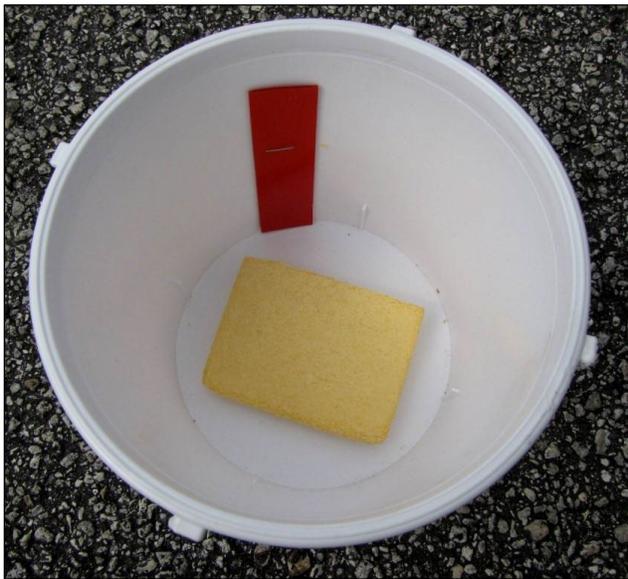
**Figures 8 and 9** Wire handle attached to trap's lid

### 3. Trap Modifications to Improve Moth Specimen Quality

There are two trap modifications that may be used to improve moth specimen quality. In regions where little or no rain is expected, a sponge can be used to soak up small quantities of water (see 3a Sponge). This method does not require modifications to the trap. In regions where there is the possibility for significant rain, holes can be drilled into the bottom of the trap to allow for water to escape (see 3b Drain holes and wire screen). Surveyors are encouraged to use whichever method provides the highest quality moth specimens in their State's unique climatic conditions.

#### 3a. Sponge

Place a dry cellulose sponge in the bottom of the trap, as shown in **Figure 10**. The sponge will absorb rainwater (except for extremely heavy amounts) that may enter the trap, keeping the moths somewhat dry. Sponges should be purchased locally. Purchase sponges without additives such as antibiotics, fragrances, etc.



**Figure 10** Cellulose sponge (yellow) and insecticidal strip (red) inside the trap

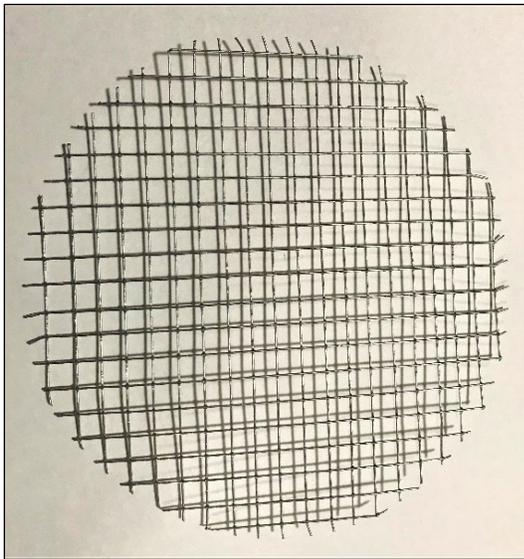


**Figure 11** Bucket with five drilled holes

#### 3b. Drain holes and wire screen

**Drain holes:** Use a  $\frac{3}{16}$ -inch drill bit to drill 4 to 5 holes in the bottom of the bucket to allow excess rainwater to drain (**Figure 11**). Holes should be drilled around the outer edge of the bucket to allow for proper drainage.

**Wire mesh screen:** Five-inch diameter wire mesh screens are available from the Survey Supply Ordering System in IPHIS (Product ID: 1381) (**Figure 12**). The wire mesh screen is placed in the bottom of the bucket trap to keep the moths from getting too wet from rainwater accumulated in the trap. When placed in the bucket trap, the screen is approximately 1-inch from the bottom of the bucket (**Figure 13**).



**Figure 12** 5 inch wire mesh screen



**Figure 13** Wire mesh screen in bucket

Moving or handling the trap may cause the screen to tilt and allow specimens to fall below the screen. To improve the stability of the wire screen mesh use four #8 x  $\frac{3}{4}$  inch machine screws placed equally distance around the trap and 1-inch from the bottom of the plastic bucket (**Tip:** Use the small tabs on the inside of the bucket as a guide for placing screws 1-inch from bottom) (**Figures 14 and 15**). The use of screws allows for the easy removal of the wire mesh screen to collect specimens that may fall below (**Figure 16**).



**Figure 14** Machine screws placed equal distance from each other around the bucket trap



**Figure 15** Machine screws shown one inch from the bottom of the bucket trap



**Figure 16** Wire mesh screen placed on machine screws inside bucket trap

#### 4. Insecticidal Strips

Insecticidal strips (**Figure 17**) are placed in traps so that captured moths are killed quickly. This step is important for preserving specimen quality. The active ingredient in the strips is Dichlorvos, also known as DDVP and Vapona. The strip may be placed in the trap by: 1) attaching it to the side of the trap (as seen in Figure 10) or 2) placing it in the bottom of the trap or on top of the screen (if using the wire mesh screen modification).



**Figure 17** Insecticidal strip

**Note:** The strip should be handled with gloves. Refer to the MSDS document for this product for safety information. Store unopened strips in a freezer that is not used for food or drink.

The CAPS program has listed a conservative length of effectiveness of 8 weeks for the insecticidal strips. The manufacturers of insecticidal strips list an effectiveness of 12 weeks. Rain, wind, high heat, and large numbers of moths may reduce the potency of the insecticidal strips. Use the 8-week interval as a starting point, but you may change the interval (not to exceed 12 weeks) based on your state's climate. If you find that moths are badly damaged (indicating they were not killed quickly), you should change the strips more frequently. In very hot and/or humid environments, it may be necessary to use two insecticidal strips, especially if traps are catching large quantities of moths.

## 5. Label the Trap

Attach a rain-proof printed label (**Figure 18**) or handwrite a note with a water-proof black marker on the bucket trap. It should indicate that the trap belongs to a state or a PPQ program. Include a phone number in case someone has concerns or questions about the trap.



**Figure 18** Label on the trap's lid



**Figure 19** Trap set in open field and away from foliage

## 6. Placement of Traps

The traps function best when placed in the open, away from foliage, as illustrated in **Figure 19**. When hung under foliage, the 3-dimensional shape of the pheromone plume (chemical in the air) is disrupted and the effectiveness of the trap is reduced. Hang the traps from places such as greenhouse roofs or in the open using metal rods (see Fig. 19) or other materials.

## 7. Trap Servicing

In the field, transfer the caught moths to labeled paper sample envelopes and store them in a cooler (Figures 20 and 21). Place them in a freezer overnight to kill any surviving specimens. Prior to screening, specimens can be stored in the freezer or in a cardboard box at room temperature.

**Note:** Do not place samples in airtight containers, like plastic bags. The moths will rot, quickly degrading specimen quality and making identification difficult or impossible.



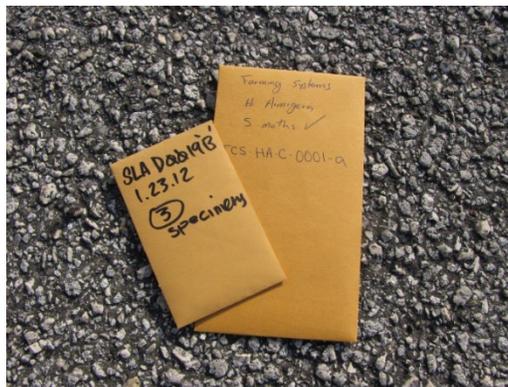
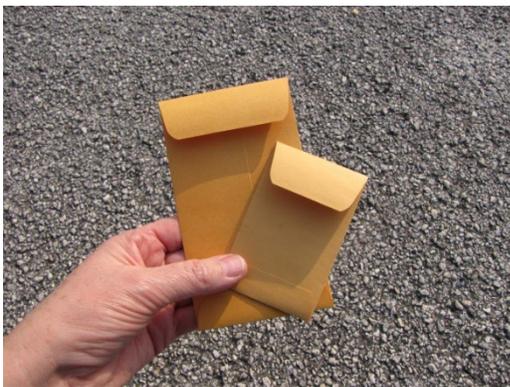
**Figure 20** Moths placed in a paper envelope



**Figure 21** Moths stored in a cooler

## 8. Sample Submission

Prior to shipping, screen the samples. Remove any moths vastly different from the target moth and all other arthropods (beetles, flies, spiders). Write the approximate number of moths being submitted on PPQ Form 391. See [Specimen Submission Guidance for Lepidoptera](#) for instructions on preparing and submitting samples.



**Figure 22** Examples of small envelopes for shipping specimens

**Tip:** The ideal envelopes for shipping samples are 3 ½ x 6 ½ inches and are commonly referred to as “coin envelopes” in office supply stores (**Figure 22**). Smaller or larger envelopes are acceptable as well.

## 9. Trap Maintenance and Storage

The general recommendation for maintenance of the plastic bucket traps is to wash them occasionally with soap and water to keep them clean, and to store them indoors, or at least protected from sun, rain, and dust. Keep the wire handle and the wire screen in good condition. The traps can be used multiple times and for multiple species since the chemicals degrade quickly in outdoor conditions. These traps usually last more than five years.

## Acknowledgments

This protocol has been designed to aid in the detection of exotic moths of concern by giving instructions on how to use generic plastic bucket traps. Photographs for figures 1-10, 17-19 and 21-22 were taken by J. Brambila; trapping materials were supplied by R. Meagher. These instructions are primarily based on work by R. Meagher.

This aid was originally prepared by Julieta Brambila, Lisa Jackson (USDA APHIS PPQ), and Robert L. Meagher (USDA ARS CMAVE) in April 2010.

This aid was revised on October 10, 2014 by Julieta Brambila, Lisa Jackson, Douglas Restom Gaskill, Andrew Derksen (USDA APHIS PPQ), and Robert L. Meagher (USDA ARS SEA). Changes include:

- In the **Pheromone** section, added an image of the septum lure. Also added images on how to secure laminate lures into lure baskets with a paper clip.
- In the **Pheromone** section, added instructions on labeling lure baskets to avoid cross-contamination from lure residue.
- In the **Trap Modification** section, noted that the sponge method may be easier and more effective than the screen method.
- In the **Insecticidal strips**, changed the recommended number of insecticidal strips to use in the trap from two to one.
- Changed the length of effectiveness of the insecticidal strips from 1 to 4 weeks to 8 weeks as a starting point; encourage states to evaluate what is effective in their climate.
- Added information on differing lengths of effectiveness of the insecticidal strips, depending on climate.
- In the **Sample submission** section, added two sample submission methods: sending moths in small envelopes or small boxes.
- In the **Sample submission** section, added images of the small manila envelopes.

This aid was revised on April 30, 2021 by Heather Moylett, Todd Gilligan, and Christopher Pierce (USDA APHIS PPQ); and reviewed by Mark Hollister (USDA APHIS PPQ). Changes include:

- In **Trap Modifications to Improve Moth Specimen Quality** section, expanded the **drain holes and wire screen** option to include how to stabilize the screen within the trap. Added figures 11-16 to show modification. Photographs taken by Christopher Pierce.
- In **Insecticidal Strips** section:
  - Removed insecticidal strip placement option: “stapling it to a string and hung inside of the trap (if large trap catches are experienced).” The two remaining placement options are simpler and effective for even large sample volumes.
  - Simplified “length of efficacy” guidance to improve clarity.
- In **Trap Servicing** section, removed plastic bags as an option for sample storage and emphasized the use of paper envelopes for sample quality. Added figure 20. Photograph taken by Todd Gilligan.
- In **Sample Submission** section, replaced written guidance with link to updated sample submission document maintained on the CAPS website.

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