Helicoverpa armigera

Scientific Name

Helicoverpa armigera (Hübner, 1808)

Synonyms:
Noctua barbara Fabricius, 1794
Noctua armigera Hübner, 1808
Heliothis peltigera var. *armigera* Ochsenheimer, 1816
Heliothis armigera (Hübner, 1808)
Heliothis conferta Walker, 1857
Heliothis pulverosa Walker, 1857
Heliothis uniformis Wallengren, 1860
Heliothis obsoleta (Fabricius, 1793)
Chloridea armigera (Hübner, 1808)
Chloridea obsoleta (Fabricius, 1793)
Helicoverpa obsoleta (Fabricius, 1793)
Heliothis rama Bhattacherjee & Gupta, 1972

Confusion between *Helicoverpa armigera* and *H. zea* exists in the older literature. Refer to Hardwick (1965) for a catalogue of literature and which species are referenced in the papers.

Figure 1. *Helicoverpa armigera* adult female from Pakistan (top), adult male from Zimbabwe (bottom) (Todd Gilligan, USDA-APHIS-PPQ-S&T).

There are three subspecies recognized: *Helicoverpa armigera armigera* (Hübner) from temperate and tropical regions of Asia, Europe and Africa, *H. a. conferta* (Walker) from Australasia, and *H. a. commoni* (Hardwick) which is confined to Canton Island in the central Pacific (Hardwick, 1965; Anderson et al., 2016, 2018).

Helicoverpa armigera and *H. zea* can interbreed under laboratory conditions producing fertile offspring, and more recently, *H. a. armigera* was found to be naturally hybridizing with *H. zea* in Brazil, resulting in individuals with varying degrees of genetic admixture (Hardwick, 1965; Laster and Hardee, 1995; Laster and Sheng, 1995; Leite et al., 2017; Anderson et al., 2018).

Common Name

Old World bollworm, scarce bordered straw worm, corn earworm, cotton bollworm, African cotton bollworm, tobacco budworm, tomato grub, tomato worm, and gram pod borer

Type of Pest

Moth, borer

Taxonomic Position

Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual CAPS Target: AHP Prioritized Pest List – 2005 through 2011, Pest of Economic and Environmental Importance – 2012 through 2019.

Pest Description

Figure 2. Newly laid eggs of *Helicoverpa armigera* (BASF Corp).

Eggs: Yellowish-white when first laid (Fig. 2), later changing to dark brown just before hatching. Eggs are gumdrop-shaped and 0.4 to 0.6 mm (<1/32 in) in diameter. The top is smooth, otherwise the surface contains approximately 24 longitudinal ribs (Bhatt and Patel, 2001; CABI, 2018).

Larvae: Larval color darkens with successive molts for the six instars typically observed for *H. armigera*. Coloration can vary considerably (Fig. 3 A, B), ranging from green, green with stripes, brown, and black (Yamasaki et al., 2009). Freshly emerged first instars are translucent and yellowish-white in color. The head, prothoracic shield, supra-anal shield and prothoracic legs are dark-brown to black as are the spiracles and raised base of the setae. The larvae have a spotted appearance (Fig. 3 A, B) due to sclerotized setae, tubercle bases, and spiracles (King, 1994; Bhatt and Patel, 2001). Second instars are yellowish green in color with black thoracic legs. Five abdominal prolegs are present on the third to sixth, and tenth abdominal segments.

The full grown larvae are highly variable and are brownish, reddish, or pale green with brown lateral stripes and a distinct dorsal stripe; larvae are long and ventrally flattened but convex dorsally. Larval size in the final instar ranges from 3.5 to 4.2 cm (approx. 1 3/8 to 1 5/8 in) in length (King, 1994).

Pupae: Pupae are dark tan to mahogany brown (Fig. 3 C), 14 to 22 mm (approx. 9/16 to 7/8 in) long, and 4.5 to 6.5 mm (approx. 3/16 to 1/4 in) wide. Body is rounded both anteriorly and posteriorly, with two tapering parallel spines at posterior tip (Hardwick, 1965).

Adults: A stout-bodied moth with typical noctuid appearance, with 3.5 to 4 cm (approx. 1 3/8 to 1 9/16 in) wing span; body is 14 to 19 mm (approx. 9/16 to 3/4 in) long. Color is variable, but the forewings in males are usually yellowish-brown, possibly patterned with grayish-green or olive-green when a fresh specimen, and sometimes marked with pink, fading to a light yellow or light brown in older specimens (Hardwick, 1965). Females are darker, usually a dull orange-brown, reddish-brown or brick red, and fading over time to a light orange-fawn or fawn (Fig. 1, 3 D) (Hardwick, 1965). Forewings have a black or dark brown kidney-shaped marking near the center (Brambila, 2009a). Hind wings are creamy white or dull yellow in both sexes with a wide dark brown or dark gray band on the outer margin (Brambila, 2009a). Identification of adult *H. armigera* requires

dissection of genitalia (Common, 1953; Kirkpatrick, 1961; Hardwick, 1965).

For more information, see Common (1953), Dominguez Garcia-Tejero (1957), Kirkpatrick (1961), Hardwick (1965, 1970), Cayrol (1972), Delattre (1973), and King (1994).

Biology and Ecology

The developmental timeline, diapause, and number of generations of *H. armigera* is regulated by the interaction between photoperiod and temperature, causing them to vary based on climate (Mironidis, 2014; Mironidis and Savopoulou-Soultani, 2012). The duration of the different life stages decreases as temperature increases from 13.3 to 32.5°C (56 to 91°F), requiring approximately 475 degree days to complete development

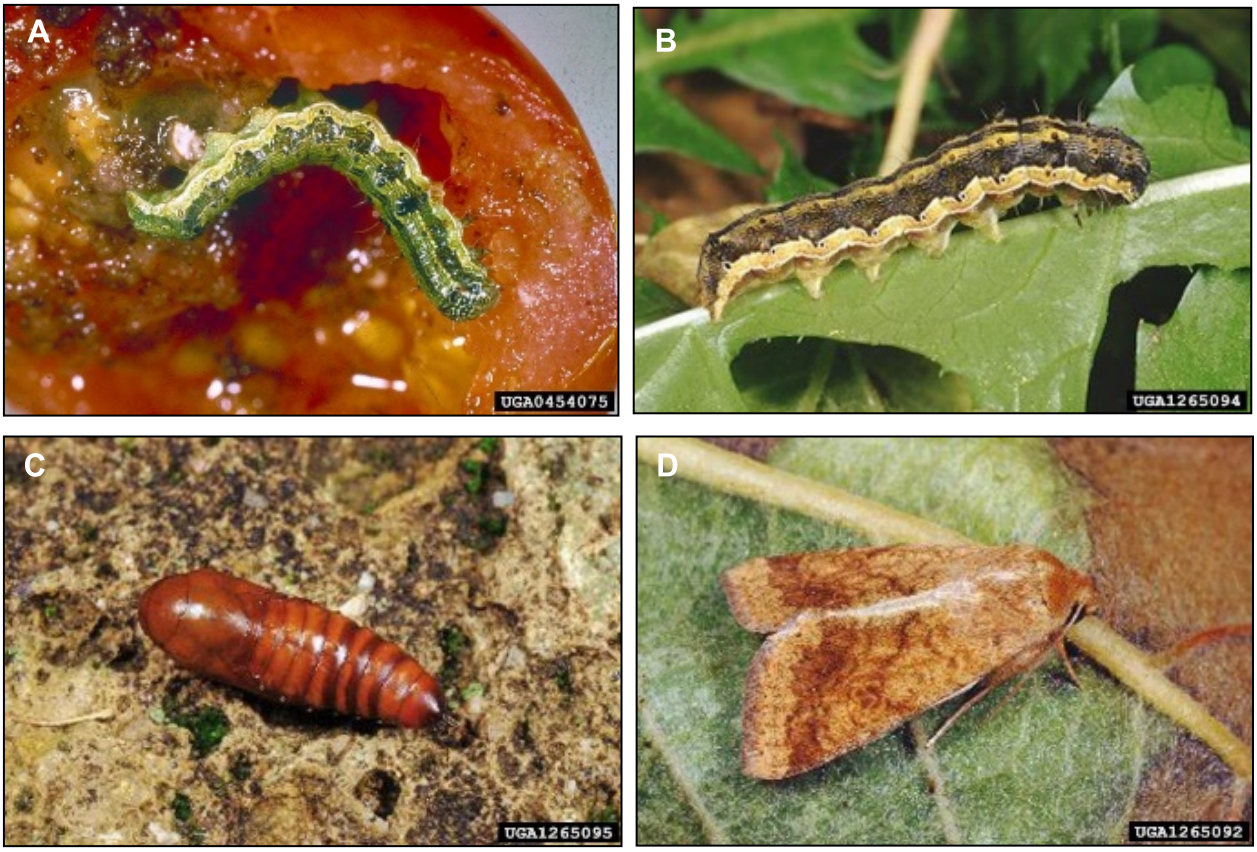

Figure 3. Life stages of *Helicoverpa armigera* (images not to scale): (A, B) larva, (C) pupa, and (D) adult. (Central Science Laboratory, Harpenden Archive, British Crown and Paolo Mazzei www.bugwood.org).

from egg to adult (Mironidis, 2014; Mironidis and Savopoulou-Soultani, 2012). In temperate regions, *H. armigera* enter into facultative winter diapause when day-length grows shorter (10 to 12 hours) and ambient temperatures drop from 24°C to 15°C (75.2 to 59°F). However, in regions or during seasons when ambient temperatures are at 25°C (77°F) or greater, few individuals enter diapause, and when temperatures are 15°C (59°F) or lower all individuals enter diapause, regardless of changes in day length (Mironidis and Savopoulou-Soultani, 2012). Additionally, when exposed to prolonged hot (≥ 37°C (98.6°F)), dry conditions larvae may enter into a summer diapause (Hackett

and Gatehouse, 1982; Nibouche, 1998). Because *H. armigera* exhibit overlapping generations, it can be difficult to determine the number of completed generations. Typically, two to five generations are achieved in subtropical and temperate regions and up to 11 generations can occur under optimal conditions, particularly in tropical areas (Tripathi and Singh, 1991; King, 1994;).

In temperate regions, *Helicoverpa armigera* overwinters in the soil in the pupal stage. Adult moths emerge May to June, depending on the latitude, and begin feeding on nectar within a few hours (Firempong and Zalucki, 1990b; DPI&F, 2005). The adults are active during the day, but most activity occurs at night starting at dusk (reviewed in Zalucki et al, 1986). Mating typically occurs for the first time on the third or fourth night after eclosion (Hardwick, 1965). They can mate several times (up to seven has been observed) prior to laying hundreds of single eggs or clusters over a period of days (Hardwick, 1965; Firempong and Zalucki, 1990b; DPI&F, 2005). A single female can lay 3,000 to 4,400 eggs under laboratory conditions, but the average in the field may be closer to 500 -1000 (Hardwick, 1965; Shanower et al, 1997; Mironidis and Savopoulou-Soultni, 2012). When selecting oviposition sites, female moths consistently prefer plants in flower (reviewed in Fitt, 1989; Firempong and Zalucki, 1990b) and tend to choose pubescent (hairy) surfaces over smooth (King, 1994). Eggs are typically laid on or near floral structures or growth points (Firempong and Zalucki, 1990b), but may be found on leaves. Duffield and Chapple (2001) found female moths prefer to lay their eggs on the underside of fully expanded leaves in the top 20 cm (8 inches) of the canopy in irrigated soybean, but preference switched to developing flowers and pods as the plants matured.

Following eclosion, first instars will consume all or part of their eggshells before moving to feed on leaf surfaces or floral structures (Hardwick, 1965; King, 1994). The early instar larva will then enter the reproductive organs, including flowers, bolls, or fruits (Hardwick, 1965; DPI&F, 2005). Later instar larvae are aggressive, often cannibalizing younger larvae when encountered, resulting in one larva per flower or fruit (as reviewed by Zalucki et al., 1986; Kakimoto et al., 2003). The number of larval instars varies from five to seven, with six being most common (Hardwick, 1965). Mature larvae drop off the host plant and pupate 2 to 17.5 cm (approx. ¾ to 7 in) below the soil surface in a silk-lined chamber, though pupation may occur within the host plant (Hardwick, 1965; DPI&F, 2005). During the growing season, individuals pupate for 10-16 days (average is 13.2 days) before emerging as adults to start the next generation (Hardwick, 1965; DPI&F, 2005).

Adult *H. armigera* can disperse distances of 10 km (6.2 mi.) during non-migratory flights and 600 to 1000 km (to 2,000 km possible; 372.8 to 621.3 mi., 1242.7 mi.) during seasonal migration (Fitt, 1989; Feng et al., 2009). Migration allows *H. armigera* to take advantage of hosts in regions that may be otherwise unsuitable for establishment (Nibouche et al., 1998; Saito, 1999; Zhou et al., 2000; Casimero et al., 2001). In China, *H. armigera* migrate northward, over the Bohai Sea, on southerly winds in the spring and summer, produce one to two generations, and then their offspring return south on northerly winds in the fall (Feng et al. 2009).

For further information, see Dominguez Garcia-Tejero (1957), Pearson (1958), Hardwick (1965), Cayrol (1972), Delattre (1973), Hackett and Gatehouse (1982), King (1994), and CABI (2018).

Damage

Helicoverpa armigera larvae prefer to feed on reproductive parts of hosts (flowers and fruits) but may also feed on foliage. Feeding damage results in holes bored into reproductive structures and feeding within the plant. It may be necessary to cut open the plant organs to detect the pest. Secondary pathogens (fungi, bacteria) may develop due to the wounding of the plant. Frass may occur alongside the feeding hole from larval feeding within.

Figure 4. Larva feeding on corn cob (Antoine Gyonnet, Lépidoptères Poitou-Charentes, www.bugwood.org).

Chickpea: Attacked from the seedling stage until maturity (Saoud, et al., 1989). Larvae (1st, 2nd, and 3rd instars) feed on the foliage (young leaves) of chickpeas, sometimes destroying seedlings completely, but preferring the flowers and flower buds of other crops at this stage (Patil et al., 2017). The highest concentration of larvae on this crop occurs during the pod formation stage (reviewed by Bajya and Monga, 2009). Larger larvae bore into pods and consume developing seed, although younger larvae may occasionally attack pods as well (Patil et al., 2017).

Corn: Eggs are laid on the silks, which are eaten by the first and second instars (Hosseininejad et al., 2015). Third to fifth instar larvae invade the ears (Fig. 4), staying hidden from natural enemies, and the developing grain is consumed (Hosseininejad et al., 2015). Larvae are absent from the plants late in the season when the stalks have dried out (Iqbal and Mohyuddin, 1990). Secondary bacterial infections occur frequently (EPPO/CABI, n.d.)

Cotton: Bore holes are visible at the base of flower buds, and the buds are hollowed out. Bracteoles are spread out and curled downwards. Leaves and shoots may also be consumed by larvae (EPPO/CABI, n.d.). "…all stages of plant growth may be attacked but reproductive tissue is preferred. Seedlings can be 'tipped out' when terminal buds are eaten. Chewing damage to squares and small bolls may cause them to shed, and chewing damage to maturing bolls may prevent normal development…" (DAF, 2018).

Peanut: The leaves, and sometimes flowers, are attacked by larvae; severe infestations cause defoliation (War et al., 2012). Peanut cultivars vary in their ability to resist attack (War et al., 2012).

Pigeon pea: Flower buds and flowers bored by small larvae and may drop; larger larvae bore into locules of pods and consume developing seed (Patil et al., 2012). Medium-

and late-maturing varieties are more susceptible to attack than early-maturing varieties (Yadava et al., 1983).

Pearl millet: Larvae attack and infest the panicle at different stages of development: flowering, milky grain, and hard grain (Singh et al., 1982, Juneja et al., 2015). The larvae feed in the open initially, but at the third instar stage they form a false web formed of excreta and dried florets which they remain underneath (Singh et al., 1982). Many larvae of different stages may be found on the same panicle simultaneously (Singh et al., 1982).

Sorghum: More than 85% of the eggs are laid on the panicles prior to flowering (Franzmann et al., 2008). After eclosion young larvae feed on anthers, switching to the developing seeds as they become available, usually around the time the larvae are in their fourth instar (Franzmann et al., 2008).

Tomato: Upon eclosion, the larvae erode the leaves, flower buds and flowers if those are available (Pinto et al., 1997). They will frequently bore into the succulent growing tip of the plant, resulting in a distorted growth pattern, however they mainly bore into the developing fruits, causing severe damage (Hardwick, 1965; Pinto et al., 1997; Mandaokar et al., 2000). Pratissoli et al. (2015) in Brazil noted that the attacked berries were medium to advanced in developmental stage, and were about 4 cm in diameter. The lesions they observed were simple holes up to large areas of destruction reaching the endocarp (Pratissoli et al., 2015). A larva can damage numerous berries prior to pupation (Pinto et al., 1997).

Pest Importance

Helicoverpa armigera is a member of the “Heliothis” clade, a subgroup of polyphagous heliothine moths, some of which are considered important pests of field and horticultural crops (Fitt, 1989; Cunningham and Zalucki, 2014). *Heliothis virescens* and the closely related *Helicoverpa zea* also belong to the “Heliothis” clade and are two key pests that are actively managed for in the United States (Cunningham and Zalucki, 2014; Kriticos et al., 2015). Established *Helicoverpa zea* populations are concentrated east of the 100th meridian and south of the 40th parallel, but seasonally migrate to northern states and Canada. *Heliothis virescens* follows a similar pattern, with populations concentrated in the eastern and southwestern United States, with annual migrations north. Based on climate suitability and crop availability, *H. armigera* geographic distribution within the United States is likely to mirror that of *H. zea*. It is not known if *H. armigera* shares the same pupal cold tolerance limits as *H. zea*. If *H. armigera* has a greater cold tolerance, established populations may not be limited to the 40th parallel (Kriticos et al., 2015) potentially increasing pest pressure on crops in northern states.

The host range of the three moths overlap considerably. This is advantageous, as the insecticides labelled for *Heliothis virescens* and *H. zea* control (e.g. Bacheler and Reisig, 2013) will likely be effective to some degree in treating *H. armigera*. However, crops that are hosts of *H. armigera* that are not common hosts for other heliothine pests; including apple, barley, Bermuda grass, carrot, kale, mango, mint, nectarine, peach, plum, allium and safflower (CABI, 2018), may require additional action to

achieve adequate control.

Based on control programs overseas, successful management is possible through employment of genetically modified (GM) crops in combination with Insecticide Resistance Management (IRM) strategies (Fitt, 2000). The United States currently grows GM *Bt* (*Bacillus thuringiensis*) corn, soybean, and cotton to control *H. zea* and *Heliothis virescens*. However, resistance to the Cry proteins expressed in first two generations of *Bt* crops developed within 7 and 13 years, respectively. Insecticide resistance is an issue for the heliothines native to the United States, the broadscale pattern of insecticide resistance is unique to *H. armigera* (Fitt, 1994; McCaffrey et al., 1989; Trowell et al., 1993; Konus et al., 2008; Kriticos et al., 2015; Wilson et al., 2018). Reproductively viable *H. armigera* x *H. zea* hybrid moths containing *H. armigera* resistance genes could potentially complicate management programs. The next generation of *Bt* cotton products, Bollgard® 3 (Monsanto), containing a Vip3A gene is now on the market, however, integrated pest management (IPM) may be the best tactic for reducing pest numbers, including proactively managing the moth year-round (Wilson et al., 2018).

Known Hosts

Note: *Helicoverpa armigera* is a widely polyphagous species attacking plants in a wide range of families, including Asterceae, Fabaceae, Malvaceae, Poaceae, and Solanaceae (Zalucki et al., 1986). The larvae will feed on at least 60 species of economically important plants (as reviewed by Fitt, 1989). Not all host plants are equally preferred for oviposition but can be utilized in the absence of a preferred host.

For a complete listing of hosts see CABI (2018) or Nibouche (1999) which has a list of 217 plants in 50 families. There have been many studies within the laboratory setting on host preference for oviposition and larval and/or pupal performance. In multiple cases the adult female has been found to preferentially oviposit on plants on which performance of the juveniles is lacking comparatively, or vice versa (see Jallow et al., 2001, for an example). Jallow and Zalucki (1996) found that oviposition was highest on corn, sorghum, and tobacco, followed by cotton varieties. Cowpea and alfalfa were the least preferred hosts for oviposition. Cotton and corn were more suitable for development and reproduction of the cotton bollworm than peanut (Hou and Sheng, 2000). Pigeon pea and corn are considered to be the most suitable hosts for this insect, when compared to sorghum, red ambadi (*Hibiscus subdariffa*), marigold, and artificial diet (Bantewad and Sarode, 2000). Tobacco, corn, and sunflower were categorized as the most preferred hosts; soybean, cotton, and alfalfa were categorized as intermediate hosts; and cabbage, pigweed, and linseed were the least preferred in an additional study (Firempong and Zalucki, 1990a).

Major hosts

Abelmoschus esculentus (okra), *Brassica oleracea botyris* (cauliflower), *Brassica oleracea capitata* (cabbage), *Brassica juncea* (mustard), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper, chilli pepper), *Carthamus tinctorius* (safflower), *Cicer arietinum* (chickpea, gram), *Citrus sinensis* (sweet orange), *Dianthus caryophyllus*

(carnation), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annuus* (common sunflower), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Papaver someniferum* (breadseed poppy), *Pennisetum glaucum* (pearl millet), *Pisum sativum* (pea), *Solanum lycopersicum* (tomato), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Trifolium alexandrinum* (Egyptian clover, berseem), *Trifolium resupinatum* (reversed clover, Persian clover), *Vigna radiata* (mung bean, green gram), and *Zea mays* (corn) (Hardwick, 1965; Moradeshaghi and Poormirza, 1976; Dhandapani and Balasubramanian, 1980; Dubey et al., 1981; Singh et al., 1982; Aslam,1988; Bilapate, 1988; Hmimina, 1988; Judal and Upadhyay, 1989; Firempong and Zalucki, 1990a; Iqbal and Mohyuddin, 1990; Bilapate et al., 1991; Bhagat and Bhalani, 1994; Vos and Frinking, 1998; Dhembare, 1999; Reddy and Subbi Reddy, 1999; Bantewad and Sarode, 2000; Jallow et al., 2001; Sujalata Devi and Singh, 2001; Yase, 2001; Karsavuran and Cetin, 2002; Singh and Battu, 2002; Chaudhari et al., 2003; Kakimoto et al., 2003; Jaglan and Saini, 2003; Balakrishnan et al., 2004; Gujar et al., 2004; Kumar et al., 2004; Sujalata and Singh, 2004; Cameron et al., 2006; Kooner et al., 2006; Banu et al., 2007; Franzmann et al., 2008; AgroAtlas, 2009; Bajya and Monga, 2009; Brijesh et al., 2009; Thanavendan and Jevarani, 2010; Arora et al., 2011; Keszthelyi et al, 2011; Hemati et al., 2012; Bisane, 2013; Javed et al., 2013; Reddy and Tangtrakulwanich, 2013; Smykal et al., 2013; Cunningham and Zalucki, 2014; Girish et al., 2014; Leite et al., 2014; Piava and Yamamoto, 2014; Gill et al., 2015; Juneja et al., 2015; Parmar et al., 2015, Grande et al., 2016; Enrique et al., 2016; Abhilasha and Shekharappa, 2017; and Patil et al., 2017).

Minor natural hosts

Allium spp. (onions, garlic, leek, etc.), *Anethum graveolens* (dill), *Antirrhinum majus* (snapdragon), *Arachis hypogaea* (peanut), *Avena sativa* (oats), *Beta vulgaris* (common beet), *Brassica oleracea gongylodes* (kohlrabi), *Brassica rapa* (turnip), *Brassica oleracea* (kale), *Brassica rapa* subsp. *pekinensis* (Chinese cabbage), *Bunium persicum* (black cumin), *Calendula officinalis* (calendula), *Callistephus chinensis* (China aster), *Canavalia insifermis* (sword bean), *Cannibus sativa* (hemp), *Chicorium intybus* (chicory), *Chrysanthemum* spp. (chrysanthemum), *Citrullus lanatus* (watermelon), *Citrus limon* (lemon), *Coffea arabica* (coffee), *Crotolaria juncea* (sunn hemp), *Cucumis melo* (muskmelon), *Cucumis sativus* (cucumber), *Cucurbita maxima* (pumpkin or winter squash), *Cuminum cyminum* (cumin), *Daucus carota* (carrot), *Eleusine coracana* (finger millet), *Foeniculum vulgare* (fennel), *Fragaria* spp. (strawberries), *Glabiolus* spp. (gladiolus), *Guizotia abyssinica* (niger), *Hordeum vulgare* (barley), *Ipomoea batatas* (sweet potato), *Lablab purpureus* (hyacinth bean), *Lactuca sativa* (lettuce), *Lathyrus odoratus* (sweet pea), *Lens medic* (lentil), *Linum usitatissimum* (flax, linseed), *Macrotyloma uniflorum* (horse gram), *Malus* spp. (apple), *Mangifera indica* (mango), *Mentha spicata* (spearmint), *Pelargonium* spp. (geranium), *Phaseolus* spp. (beans), *Phaseolus vulgaris* (common bean), *Psophocarpus tetragonolobus* (winged bean), *Pyrus sativus* (pear), *Raphanus sativus* (radish), *Rosa x damascena* (damask rose), *Salvia sclarea* (clary sage), *Sambucus nigra* (elderberry), *Sesamum indicum* (sesame), *Spinacea oleracea* (spinach), *Tagetes* spp. (marigold), *Trachyspermum ammi* (carom, ajwain), *Trigoniella foenumgraecum* (fenugreek), *Triticum aestivum* (wheat), *Vicia faba* (broad bean), *Vigna mungo* (blackgram), *Vigna umbellata* (rice bean), and *Vigna unguiculata* (cowpea) (May, 1949; Hardwick, 1965; Aslam, 1988; Judal and Upadhyay, 1989; Iqbal and Mohyuddin, 1990; Bhagat and Bhalani, 1994; Shi et al., 1995; Sharma et al., 1998; Sreenivasa Rao and Koteswara Rao, 1999; Bantewad and Sarode, 2000; Sujalata Devi and

Singh, 2001; Pallavi et al., 2002; Dömötör, 2003; Kakimoto et al., 2003; Kumar et al., 2004; Bharati et al., 2007; Lin et al., 2007; Midgley et al., 2008; Bajya and Monga, 2009; Nadda et al., 2012; War et al., 2012; Cunningham and Zalucki, 2014; Leite et al., 2014; Namin et al., 2014; Manjula et al., 2015; and Golparvar and Naseri, 2016).

Poor hosts
Asparagus officinalis (asparagus)*, Azadirachta indica* (neem)*, Oryza sativa* (rice)*,* and *Vitis vinifera* (grape) (Barrion and Litsinger, 1987; Vörös, 1996; Ma et al., 2000; De Villiers and Pringle, 2007; Jha et al., 2014).

Wild hosts
Acalypha spp. (copperleaf), *Aerva sanguinolenta* (karadia), Aeschynomene indica (Indian jointvetch), *Amaranthus spinosus* (spiny amaranth), *Amaranthus* spp. (pigweed, amaranth), *Blandfurdia grandiflora* (Christmas bells), *Brassica nigra* (mustard), *Calendula arvensis* (field marigold), *Centella asiatica* (Indian penny wort), *Chenopodium album* (lambsquarters), *Chenopodium quinoa* (quinoa), *Datura metel* (datura), *Datura* spp., *Godetia grandiflora* (evening primrose), *Gomphrena* spp., *Hyoscyamus niger* (black henbane), *Malvastrum americanum* (Indian Valley false mallow), *Melanthera nivea* (snow squarestem)*, Physalis peruviana* (cape gooseberry), *Plectranthus neochilus* (boldo-rasteiro), *Ricinus communis* (castor bean), *Rumex dentatus* (toothed dock), *Rumex maritimus* (golden dock), Sesbania sesban (Egyptian riverhemp, common sesban), *Solanum nigrum* (black nightshade), *Sonchus oleraceus* (annual sowthistle), *Sphaeranthus indicus* (East Indian globe thistle), *Stellaria media* (chick weed), and *Xanthium strumarium* (common cocklebur) (Hardwick, 1965; Kraemer, 1966; Iqbal and Mohyuddin, 1990; Coombs and Ramsey, 1991; Mehta et al., 1996; Gu and Walter, 1999; Sujalata Devi and Singh, 2001; Kumar et al., 2004; CABI, 2018; Cunningham and Zalucki, 2014; and Krinski and Godoy, 2015).

Pathogens or Associated Organisms Vectored
Helicoverpa armigera is not a known vector and does not have any associated organisms.

Known Distribution
Africa: Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Cote d'Ivoire, Democratic Republic of the Congo, Egypt, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Lesotho, Libya, Madagascar, Malawi, Mali, Mauritania, Mauritius, Mayotte, Morocco, Mozambique, Namibia, Niger, Nigeria, Republic of the Congo, Réunion, Rwanda, Saint Helena, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zambia, and Zimbabwe; **Asia:** Afghanistan, Armenia, Azerbaijan, Bangladesh, Bhutan, Bismarck Archipelago, Brunei, Cambodia, China, Cocos Islands, Georgia, Hong Kong, India, Indonesia, Iran, Iraq, Israel, Japan, Jordan, Kazakhstan, South Korea, Kuwait, Kyrgyzstan, Laos, Lebanon, Malaysia, Myanmar, Nepal, Pakistan, Philippines, Saudi Arabia, Singapore, Sri Lanka, Syria, Taiwan, Tajikistan, Thailand, Turkey, Turkmenistan, United Arab Emirates, Uzbekistan, Vietnam, and Yemen; **Caribbean:** Dominican Republic and Puerto Rico; **Europe:** Albania,

Andorra, Austria, Azores Islands, Balearic Islands, Belgium, Bosnia and Herzegovina, Bulgaria, Canary Islands, Corsica, Cyprus, Dodecanese Islands, Finland, France, Germany, Gibraltar, Greece, Hungary, Italy, Kriti (Crete), Lithuania, Macedonia, Madeira Island, Malta, Moldova, Montenegro, Portugal, Romania, Russia, Sardinia, Selvagens Islands, Serbia, Sicily, Slovenia, Spain, Sweden, Switzerland, the Netherlands, Turkey (European), and Ukraine; **Oceania:** American Samoa, Australia, Belau, Christmas Island, Cook Islands, Federated States of Micronesia, Fiji, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Norfolk Island, Northern Mariana Islands, Palau, Papua New Guinea, Samoa, Solomon Islands, Tonga, Tuvalu, and Vanuatu; **South America:** Argentina, Bolivia, Brazil, Colombia, Paraguay, Peru, Suriname, and Uruguay (Kazimierczak, 2009; Ugurlu, S. 2009; Fibiger and Skule, 2011; Radonjić and Hrnčić, 2011; EPPO, 2009; Czepak et al, 2013; GPDD, 2013; Keszthelyi et al., 2013; Sugayama, 2013; Senave, 2013; Specht et al., 2013; Tay et al., 2013; de Jong et al., 2014; Leite et al., 2014; Murúa et al., 2014; NAPPO, 2014; Castiglioni et al., 2016; CABI, 2018).

In October 2012 a female moth was caught in a cargo facility in Michigan, and in June of 2015 one male moth, and in early July of 2015, three adult male moths were found in Manatee County, Florida (PPQ, 2014; FDACS, 2015; NAPPO, 2016). Subsequent intensive and extensive surveying throughout the state of Florida for a complete year yielded no additional sightings, so the incident was deemed an isolated incident. (NAPPO, 2016). Continued trapping efforts throughout 2017 and early 2018 have likewise not yielded any positive sightings (CERIS, 2018). It is considered “Absent, no longer present at this time” within the continental United States.

It was previously recorded in Croatia, Czech Republic, Estonia, Latvia, Norway, Poland, Slovakia, and in Great Britain (including the Channel Islands, England, Northern Ireland, and Wales) but the bollworm is considered eradicated or otherwise not present in those countries (GPDD, 2013).

Pathway

Helicoverpa armigera could potentially move through international trade (Lammers and MacLeod, 2007). This species has been intercepted over 1,300 times at U.S. ports of entry (Pest ID, 2018). Most material was for consumption (1,263) while the rest was for non-entry (37) and propagation (4). Plant material interceptions have occurred on: *Tagetes* sp. (82), *Bupleurum* sp. (78), *Ornithogalum* sp. (71), *Leucospermum* sp. (62), *Capsicum* sp. (56), *Veronica* sp. (50), and *Cicer arietinum* (chickpea) (31), among many others. Most interceptions originated on material from the Netherlands (310), Israel (241), India (171), Kenya (57), Italy (43), Spain (33), and Zimbabwe (32) (Pest ID, 2018). Additionally, 9,420 interceptions were recorded of Helicoverpa spp. of which many could be *armigera* as well (Pest ID, 2018).

In 2013, *H. armigera* was confirmed to be established in Brazil. It has since spread through South America and was detected in Puerto Rico in 2015 (reviewed in Kriticos et al., 2015). With its recent establishment in the new world, natural spread through migration or “land-hopping” from Central America or the Caribbean is considered a likely pathway into North America (Kriticos et al., 2015).

Potential Distribution within the United States

As stated previously, according to Venette et al. (2003) approximately 49% of the continental U.S. would be suitable habitat for the pest based on climate zones. According to their model, the area at risk in the west is somewhat patchy: all of Nevada; most of Utah, Arizona and New Mexico; and parts of Washington, Oregon, California, Idaho, Wyoming, and Colorado. Texas is also partially at risk. The eastern states at risk form a large contiguous swath up to Maine from Texas eastward, and also include states west of Lake Michigan including Minnesota and Wisconsin.

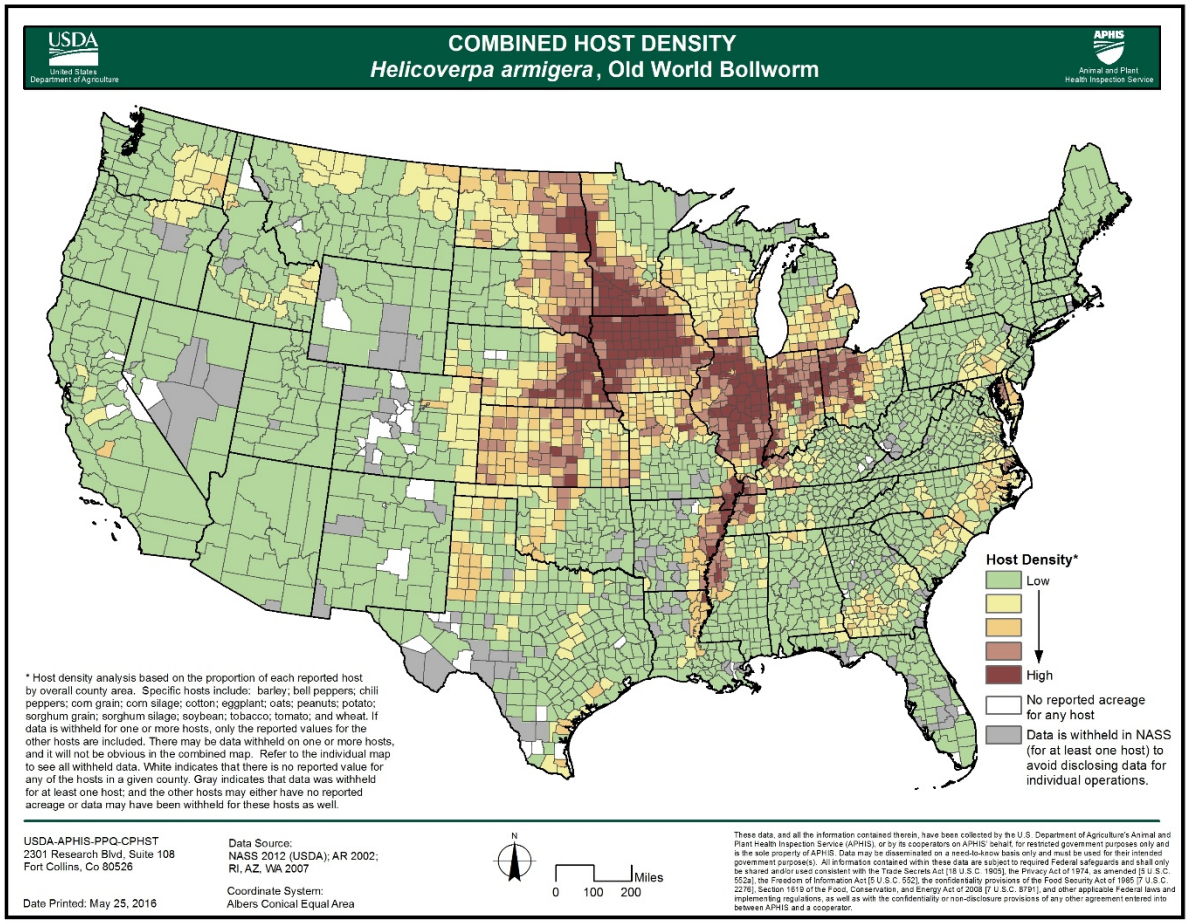

Figure 5. Combined host density map for the continental United States. Crop density for counties where crops reported as grown in 2012. The specific crops are barley, bell peppers, chili peppers, corn (grain and silage), cotton, eggplant, oats, peanuts, potato, sorghum (grain and silage), soybean, tobacco, tomato and wheat. Courtesy of USDA-APHIS-PPQ-CPHST Fort Collins.

In 2014, *H. armigera* was detected in Puerto Rico. The pest is not known to be established in the conterminous United States.

Survey

Approved Methods for Pest Surveillance*:

The CAPS-approved method is a trap and lure combination. For negative data reporting, use the approved lure: *Helicoverpa armigera* Lure with one of the approved traps: 1) plastic bucket trap or 2) heliothus trap.

The lure is effective for 28 days (4 weeks). The length of effectiveness of this lure may be reduced in hot and dry climates. In these environments, lures may need to be changed every two weeks instead of every four weeks.

The Plastic Bucket Trap is also known as the unitrap. The trap has a green canopy, yellow funnel, and white bucket and is used with a dry kill strip (Fig. 6). See the Plastic Bucket Trap Protocol (Brambila et al., 2014) for instructions on using the plastic bucket trap.

The Texas (Hartstack) trap is not available commercially. See Hartstack et al. (1979) or Johnson and McNeil (no date) for images and trap design.

Figure 6. Old world bollworm trap. (USDA-APHIS, Plant Protection and Quarantine).

The product names in the IPHIS Survey Supply Ordering System:

1) Plastic Bucket Trap
2) Heliothis Trap
3) *Helicoverpa armigera* Lure

IMPORTANT: Do not include lures for other target species in the trap when trapping for this target.

Trap spacing: When trapping for more than one species of moth (that require different lures), separate traps for different moth species by at least 20 meters (65 feet).

Survey site selection:
This species can be surveyed for in a variety of crops due to its polyphagous nature. The larvae feed mainly on the flowers and fruit of the crops. *Helicoverpa armigera* is known to infest high value crops, including tomatoes, cotton, and corn.

Trap placement:
Traps should be placed 1.5 to 1.8 m (~5 to 6 ft) above the ground (Aheer et al., 2009; Kant et al., 1999; and Zhou et al., 2000).

Time of year to survey:
Adult moths emerge between April and June depending on latitude, and can be observed until October, because of the long migration period. Moths emerge in May to June depending on latitude, and lay eggs singly on a variety of host plants on or near floral structures.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at https://caps.ceris.purdue.edu/.

Literature-Based Methods:

Trapping: (From Venette et al., 2003). Pheromone traps using (*Z*)-11-hexadecenal and (*Z*)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera* (Pawar et al., 1988; Loganathan and Uthamasamy, 1998; Loganathan et al., 1999; Visalakshmi et al., 2000; Zhou et al., 2000). Of three pheromone doses tested in the field (0.75, 1.0, and 1.25 mg/septum), 1 mg attracted the most males (Loganathan and Uthamasamy, 1998); the trap type was not specified. Rubber septa impregnated with these sex pheromone components (1 mg/septum) were equally effective in capturing males for 11 days in the laboratory (Loganathan et al., 1999). Captures of *H. armigera* in the field were significantly lower with 15-day-old lures than with fresh lures, and the authors recommend replacing lures every 13 days (Loganathan et al., 1999). Pawar et al. (1988) made similar observations, but reported that the pheromone lasted up to 40 days. Males responded to the pheromone during dark hours only, commencing at 6:00 PM and terminating at 6:00 AM. The highest response was between 11:00 PM and 4:00 AM (Kant et al., 1999). When trapping the moth in pigeonpea, Shanower et al. (1999, as reviewed by Dayalal et al. (2015)) found that 50 traps per hectare was better than 30 or 40 traps per hectare for trapping larger numbers of male moths, and the resulting population of eggs and larvae and percent pod damage were statistically significantly lower.

Trap design has a significant impact on the number of male *H. armigera* moths that will be captured with pheromone lures. Funnel traps and Texas (Hartstack) traps are substantially more effective than sticky traps (Kant et al., 1999) and cone traps are significantly more effective than water-pan traps (Sheng et al., 2002).

Not recommended:

Visual inspections of plants for eggs and/or larvae are frequently used to monitor and assess population sizes for *H. armigera*. However, this approach can be time consuming and unreliable. Females lay several hundred eggs on a variety of host plants (Duffield and Chapple, 2001). Eggs may be difficult to detect. The eggs are laid singly, often on the underside of leaves, and hatch in less than three days at an optimum temperature of 27 to 28°C (81 to 82°F). While feeding, larvae may be seen on the surface of plants, but they are often hidden within the fruit or flower. Bore holes and heaps of frass (excrement) may be visible, but otherwise it is necessary to cut open the plant organs, especially damaged fruit, to detect the pest (Bouchard et al., 1992).

In vegetative Australian cotton and irrigated soybean, a minimum of 60 whole plants per 100 hectare commercial field are examined for the presence of *H. armigera* eggs or larvae. Only the upper terminal (approximately 20 cm or 8 in) of a plant is inspected when cotton plants begin to produce squares, or on the undersides of leaves during early development in soybeans, and then on developing flowers and pods as they mature (Brown, 1984; Dillon and Fitt, 1995; Duffield and Chapple, 2001). In experimental plots, visual inspections for *H. armigera* in pigeon pea were restricted to the upper third of whole plants (four sets of five plants in a 30 x 30 meter plot) (Sigsgaard and Ersbøll, 1999).

Leaves of tomato plants are more attractive than flowers or fruits as *H. armigera* oviposition sites, but use of a single-leaf sample unit (with a sample size of 30 plants per field) has proven ineffective in detecting low densities of *H. armigera* (Cameron et al., 2001). On some tomato cultivars, leaves in the upper half of the plant are preferentially selected for oviposition (Saour and Causse, 1993).

For CAPS surveys, visual survey is not an approved method for this species.

Adults of both sexes can be captured in black light traps. For CAPS surveys, light traps are not an approved method for this species as they are not species-specific.

Key Diagnostics/Identification

Approved Methods for Pest Surveillance*:

Morphological:

Confirmation of *Helicoverpa armigera* is by morphological examination. *Helicoverpa armigera* and the native, abundant species, *Helicoverpa zea,* are very similar in appearance. *Helicoverpa armigera* cannot be visually distinguished from *H. zea*; all specimens require dissection. Final identification requires dissection of adult male genitalic structures. Instructions for preparing and dissecting the specimens are available at Brambila (2009b); see below for link.

For field level screening, use:
Brambila, J. 2009a. *Helicoverpa armigera* - Old World Bollworm, Field Screening Aid and Diagnostic Aid.

Instructions for dissecting *H. armigera* are available at:
Brambila, J. 2009b. Dissection instructions for identifying male *Helicoverpa amigera* and *H. zea*.

A guide to larval identification is available at:
Passoa, S. 2007. Identification guide to larval Heliothinae (Lepidoptera: Noctuidae) of quarantine significance.

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at https://caps.ceris.purdue.edu/.

Literature-Based Methods:

Molecular:

As early as 1997, multiple molecular methodologies had been developed to differentiate Heliothinae species (see review by Arneodo et al., 2015). In the last decade, mitochrondial DNA (mtDNA) cytochrome oxidase I (COI) and sometimes cytochrome-B genes (Cyt*b*-Harm01 and Cyt*b*-Harm08, for example) have been used to differentiate *H. armigera* into haplotypes, to confirm positive identification of the species, and for confirming new host plant records (Behere et al., 2007, 2008; Tay et al., 2013; Arneodo et al., 2015; Krinski and Godoy, 2015). More recently, in 2016, Nagoshi et al. used the COI gene in combination with segments of the Z-linked triosephosphate isomerase (*Tpi*)

genes for distinguishing *H. zea* and *H. armigera*, and to identify possible hybridization.

In 2017, Zink et al. used a new technique, droplet digital PCR (ddPCR), to confirm the presence of *H. armigera* in a sample containing large quantities of *H. zea*. This third-generation PCR technique is efficient and scalable, enabling rapid detection of a single *H. armigera* leg combined with 999 *H. zea* legs by partitioning the sample into 20,000 nanoliter-sized water-in-oil sub-samples. They used an intercalating DNA dye (blue=positive), and since no probes are needed with the dye, costs are reduced as well.

There is currently no CAPS approved molecular diagnostic method for this species. Guidance for screening OWB samples using real-time PCR has been developed by S&T scientists. This diagnostic method will be considered for inclusion as a CAPS approved method once the necessary labs have passed the OWB Proficiency Test and have been certified as a molecular screening lab by USDA APHIS PPQ.

Easily Confused Species

As of 2018, there are over 40 described species of *Helicoverpa* moths (Myers et al., 2018). Several of these noctuid pests as well as others can be confused easily with *H. armigera*, including *H. assulta* and *H. punctigera* (both are not known in the United States), as well as *H. zea* and *Chloridea virescens* (formerly *Heliothis virescens*) which are both are present in the United States (Kirkpatrick, 1961; CABI, 2018). An older morphological study and key of the adults for *H. assulta, H. punctigera,* and *Chloridea virescens* is available from Kirkpatrick (1961). The adults can be identified by their genitalia and legs, including the shape of the valvae, the shape of the vesica (lobes and coils), spines on the vesica, spurs or scales on the legs, and the shape or structure of the appendix bursae in female moths (Hardwick, 1965). Specifically, spurs or scales on the legs can be used to identify the genus *Helicoverpa*, and then *armigera* males can be separated from *H. zea* males by the lobes at the base of their vesica (1 vs. 3), and the number of coils of the vesica (6.5-8.5 vs. 8.0-11.0), while the females can be determined by the spicules (tiny spines) on the surface of the appendix bursae which are mostly absent in *H. zea* (Hardwick, 1965, Pogue, 2004).

The early instar larvae can be difficult or impossible to identify to species, but can be separated from other noctuid genera by their “spiny” cuticle/skin (Hardwick, 1965). Larvae of *H. armigera* and *H. zea* must be separated from each other using molecular methods, as morphological methods are inadequate (Gilligan and Passoa, 2014; Gilligan et al., 2015). Cahill et al. (1984) provide morphological information to distinguish third/fourth and sixth instars of *H. armigera* and *H. punctigera*. Use Brambila (2009a) and Brambila (2009b) to screen for adult *H. armigera* males.

Commonly Encountered Non-targets

The native species *Helicoverpa zea* is strongly attracted to the *H. armigera* pheromone lure. Differentiation between *H. armigera* and *H. zea* is very difficult; identification is by dissection of internal structures of adult males (Pogue, 2004).

In addition, some native *Spodoptera* species frequently occur in *H. armigera* traps, including male and female *Spodoptera frugiperda* and *S. ornithogalli*. To the untrained observer, these moths may look similar to the target (all are brownish colored moths); however, on closer inspection, the *Spodoptera* moths can be screened out of the samples. *Spodoptera frugiperda* is smaller, with narrower wings, and tends to be grey. *Spodoptera ornithogalli* is similar in size, but its wings are banded cream and dark brown.

Figure 7. *Leucania adjuta*. Photo courtesy of Mark J. Dreiling.

Another species that is commonly found in *H. armigera* traps is *Leucania adjuta* (J. Brambila, personal communication, 2014). This non-target moth may occur in large numbers in traps. *Leucania adjuta* males (Fig. 7) are generally similar in size and color to *Helicoverpa zea* and *H. armigera* but have various differences in wing color patterns (Brambila, personal comm., 2014).

Surveyors should screen these moths out if possible; however, the specimens may be submitted if the moths are in poor condition or the surveyor does not feel comfortable screening these non-target out of the traps.

For images of genitalia of the native moth, *Leucania adjuta* see: Brambila, J. 2010. Images of *Leucania adjuta* genitalia.

For additional images of *Leucania adjuta*, see:

http://www.nearctica.com/leucania/sysfly/Ladjuta.htm

http://mothphotographersgroup.msstate.edu/species.php?hodges=10456

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Revisions

April 2014

1) Revised the **Key Diagnostics/Identification** section.
2) Revised the **Easily Confused Species** section.
3) Added the **Commonly Encountered Non-targets** section.
4) Added Figure 6 and link to Brambila, J. 2010. Images of *Leucania adjuta* genitalia.

Last update: June 2018

July 2014
1) Revised the **Distribution** section.

June 2018
1) Revised the **Synonyms**
2) Revised the **Reason for Inclusion in Manual**
3) Revised the **Pest Description** section
4) Revised the **Biology and Ecology** section
5) Revised the **Damage** section
6) Revised the **Pest Importance** section
7) Revised the **Known Hosts** section
8) Revised the **Known Distribution** section
9) Revised the **Pathway** section
10) Revised the **Potential Distribution within the United States** section
11) Revised the Literature-Based Methods in the **Survey** section
12) Revised the Literature-Based Methods in the **Key Diagnostics/Identification** section
13) Added **References**