

Biology and Management of the Whitemarked Tussock Moth (Lepidoptera: Erebidae)

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Abstract

The whitemarked tussock moth, *Orygia leucostigma* J.E. Smith (Lepidoptera: Erebidae), is common in the eastern United States and Canada but occurs as far west as Alberta, Colorado, and Texas. Larvae are conspicuously colored, with distinctive red heads, white or yellow mid-dorsal tufts of hairs, and a black mid-dorsal stripe flanked by yellow subdorsal stripes. Larvae feed on at least 140 species of deciduous and coniferous woody plants, demonstrating tolerance to a wide variety of plant defensive compounds. This species is not a major forest pest but does exhibit periodic outbreaks that can cause serious defoliation. However, it often is a pest of shade trees, walnut orchards, poplar and Christmas tree plantations, and blueberry crops, and its urticating larval hairs and setae can cause dermatitis and allergic reactions in sensitive individuals. This species is likely to become more important in a warmer climate, warranting greater attention to its population dynamics and control options. Variation in temperature, host condition, and mortality agents appear to be responsible for changes in abundance. Maintaining tree vigor and planting resistant varieties are nonchemical options for management. When control is necessary, application of microbial pathogens, such as *Bacillus thuringiensis*, is effective and favored over synthetic insecticides. Other biorational insecticides include azadirachtin (a botanical insecticide) and insecticidal soap. Conventional synthetic insecticides include several pyrethroids (such as bifenthrin and permethrin), organophosphates (such as acephate and malathion), and carbamates (carbaryl). These synthetic chemicals have broad nontarget effects that discourage use in forests. Insecticides should only be used according to their label directions.

Key words: *Orygia leucostigma*, forest management, defoliator, biological control, pheromone

The whitemarked tussock moth, *Orygia leucostigma* J.E. Smith (Lepidoptera: Erebidae: Lymantriinae), is common in fields, woodlands, and forests of the eastern United States and Canada but occurs as far west as Alberta, Colorado, and Texas (Furniss and Carolin 1992, Wagner 2005). Larvae feed on at least 140 species of woody host species, including virtually all woody tree and understory species in eastern forests (Drooz 1985, Johnson and Lyon 1988, Furniss and Carolin 1992, Wagner 2005, Heppner 2007). This species is closely related to several other forest pests in the subfamily Lymantriinae, including the Douglas-fir tussock moth, *Orygia pseudotsugata* (McDunnough) (Lepidoptera: Erebidae), in the western United States, gypsy moth, *Lymantria dispar* (L.) (Lepidoptera: Erebidae), introduced to the United States from Eurasia, and the nun moth, *Lymantria monacha* (L.) (Lepidoptera: Erebidae), a Eurasian species that could become invasive if introduced to the United States.

Although the whitemarked tussock moth is considered to be a minor pest in forests in the United States (Drooz 1985, Furniss and Carolin 1992), outbreaks in the northern United States and Canada can severely defoliate trees over large areas (Webster 1916,

Wilson 1991, van Frankenhuyzen et al. 2002, Grant et al. 2003). One year of severe defoliation can kill balsam fir, *Abies balsamea* L. (Pinales: Pinaceae) (van Frankenhuyzen et al. 2002). However, this species more often is a serious pest of shade or ornamental trees, black walnut, *Juglans nigra* L. (Fagales: Juglandaceae), and Christmas tree plantations, and blueberries, *Vaccinium corymbosum* L. (Ericales: Ericaceae) (Drooz 1985, Furniss and Carolin 1992, van Frankenhuyzen et al. 2002, Wagner 2005, Isaacs and van Timmeren 2009, Hall and Buss 2014, Hyche 2018). Larvae can occur at sufficient densities to cause severe defoliation of shade and ornamental trees (Hall and Buss 2014, Hyche 2018). Frass and hairs frequently are nuisances and potential health threats for homeowners in urban areas, where the hairs on the larvae or in cocoons cause dermatitis and allergic reactions in sensitive individuals (Knight 1922; Gilmer 1925; Goldman et al. 1960; Diaz 2005; Wagner 2005; Hossler 2009, 2010; Hyche 2018). Hairs in cocoons retain their urticating capability for a year or more (Hall and Buss 2014).

Populations typically are regulated by a variety of predators, parasitoids, and entomopathogens and are controlled by

insecticide applications recommended for other defoliators (Isaacs and van Timmeren 2012, Hall and Buss 2014, Louisiana Insect Pest Management Guide 2018). When necessary, control for whitemarked tussock moth can be achieved with commercially available microbial insecticides. This profile is intended to provide extension specialists, forest managers, and public health providers with an overview of factors affecting population dynamics, public health issues, and control options for the whitemarked tussock moth.

Description

Male moths are ash gray with a wingspan of 26–30 mm (1–1.2") and prominent bipectinate antennae (Fig. 1). The forewing has a conspicuous white spot near the anal angle and is marked by wavy bands (Fig. 1).

Females are gray, hairy, about 12 mm (0.5") long, and brachypterous (reduced wings) (Fig. 2; Drooz 1985, Furniss and Carolin 1992, Hyche 2018). Females cannot fly but attract males to their cocoons upon emerging (Grant et al. 2003, 2006) and lay 150–200 eggs in a frothy mass on the surface of the cocoon (Fig. 3; Johnson and Lyon 1988, Thurston and MacGregor 2003, Wagner 2005). Tammaru et al. (2002) and Thurston and MacGregor (2003) reported that fecundity increases linearly with female size (pupal weight or volume, respectively). Females die after ovipositing and fall to the ground.

Mature larvae are 25–37 mm long (1–1.5") and are conspicuously colored, with distinctive red heads, white or yellow mid-dorsal tufts of hairs, and a black mid-dorsal stripe flanked by yellow sub-dorsal stripes (Fig. 4; Godfrey 1987, Wagner 2005, Hyche 2018). They also have paired hair pencils of black setae that extend forward from the prothorax and another hair pencil extending backward from the eighth abdominal segment and red dorsal glandular structures on segments six and seven (Godfrey 1987, Hall and Buss 2014, Hyche 2018).

Larvae often are seen wandering on understory vegetation, on the walls of structures, and on the ground prior to pupation. Cream- or

tan-colored cocoons are spun from silk and some setae in protected sites in bark crevices (Fig. 5), tree cavities, and under siding or soffits of buildings (Webster 1916, Wilson 1991, Hall and Buss 2014).

Ecology

Phenology and Life Cycle

This species overwinters in the egg stage, and larvae emerge from April to June, depending on location (Johnson and Lyon 1988, Wagner 2005). Newly emerged larvae often disperse by ballooning, offsetting the disadvantage of flightless females (Thurston and MacGregor 2003). Young larvae feed on the surface of leaves, causing skeletonization. As they mature, they chew holes in leaves and eventually consume all but the major veins (Drooz 1985, Johnson and Lyon 1988). Larvae mature in 5–6 wk, and then pupate for about 2 wk. Generally, this species has two, or more, generations per year (Drooz 1985, Wagner 2005, Reynolds et al. 2007).

Larval development rate is related to temperature. Isaacs and van Timmeren (2009) reported that larval developmental rate increased significantly with temperature from 16°C (61°F) to 28°C (82°F), but development ceased below 10°C (50°F) and above 35°C (95°F). Reynolds et al. (2007) noted that in the northeastern United States, the whitemarked tussock moth is facultatively bivoltine in warm years and univoltine in cooler years, suggesting that warming temperatures could double population growth rate.

Upon emerging from their cocoons, flightless females attract males through pheromone communication. Grant et al. (2003)



Fig. 1. Adult male whitemarked tussock moth. Note the distinctive white spot near the anal angle of the front wing. Photo by Mark Dreiling, courtesy of Bugwood.org.

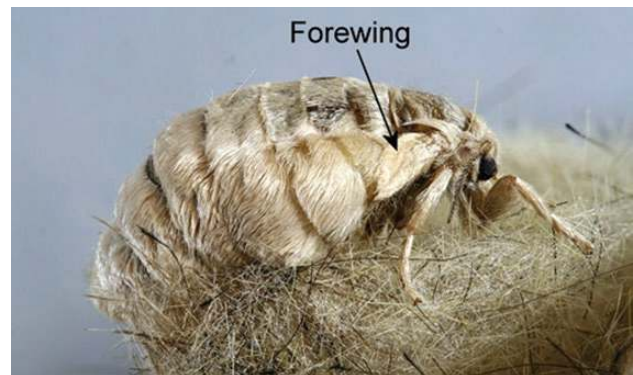


Fig. 2. Adult female whitemarked tussock moth, showing reduced wings. Photo by Donald W. Hall, University of Florida, with permission.



Fig. 3. Adult female whitemarked tussock moth with egg mass on cocoon. Photo by John L. Foltz, University of Florida, courtesy of Bugwood.org.



Fig. 4. Mature whitemarked tussock moth caterpillar. Photo by A. Steven Munson, USDA Forest Service, courtesy of Bugwood.org.



Fig. 5. Whitemarked tussock moth cocoons in bark crevices. Photo by North Carolina Forest Service, courtesy of Bugwood.org.

identified (Z,Z)-6,9-heneicosadien-11-one as the only essential component of the female pheromone. Male copulatory behavior is stimulated by tarsal contact with the female's body scales (Grant 1981), which contain a series of *n*-alkanes from C-21 to C-29 that are responsible for eliciting this response (Grant et al. 1987).

Factors Affecting Population Dynamics

Whitemarked tussock moth populations fluctuate as a result of variable environmental conditions. Outbreaks were recorded as early

as 1916 (Webster 1916) and have occurred at about 15-yr intervals in the northern United States and Canada since the 1960s (van Frankenhuyzen et al. 2002). However, the extent to which this species shows a cyclic (regular period) or irruptive (irregular period) outbreak pattern has not been established. Given its relatively high thermal tolerance range (Isaacs and van Timmeren 2009), this species is likely to become more abundant as a result of a warmer climate (Reynolds et al. 2007). Variation in availability of suitable food and in abundance of predators and parasites drives population dynamics (Hall and Buss 2014, Schowalter 2016).

Food Availability

Whitemarked tussock moth is a dietary generalist, feeding on at least 140 known native plant species, including virtually all deciduous and coniferous tree and understory species in eastern forests (Erelli et al. 1998, Wagner 2005, Heppner 2007). This diversity of host species suggests exposure to a wide variety of host toxins. Karowe (1992) compared larval growth rates on an artificial control diet and nine artificial diets, each with a single, added, naturally occurring plant defensive compound, i.e., five alkaloid treatments (berberine, nicotine, caffeine, quinine, and scopolamine), two phenolic treatments (quercetin and rutin), one terpenoid treatment (α -pinene), and coumarin, at concentrations sufficient to reduce larval growth but not cause substantial mortality. He found that adaptation to one plant defensive compound was not negatively associated with adaptation to other defensive compounds, even among chemical classes, indicating little fitness trade-off among plant hosts. Although tussock moths apparently concentrate and egest tannins without metabolizing them (Barbehenn and Martin 1992, Kopper et al. 2002), terpenoids apparently are metabolized, with little effect on larval performance (Raffa and Powell 2004).

Erelli et al. (1998) reported that larval consumption rates on six tree species tended to be highest on foliage with low nitrogen concentration and that growth rates of first instars were significantly related to the interaction of nitrogen and tannin concentrations. Foliage nitrogen and tannin concentrations were significantly negatively related. Glynn et al. (2003) and Hale et al. (2005) also found that increasing nutrient availability to host black poplar, *Populus nigra* L. (Malpighiales: Salicaceae), significantly reduced production of phenolic glycosides and increased growth of larvae.

Barbehenn and Martin (1992) and Barbehenn et al. (2003) reported that tussock moth larvae are relatively tolerant of plant phenolics and show relatively little phenolic oxidation (which can damage gut epithelial tissues) in their midguts. Most ingested tannin is concentrated and egested without being metabolized (Barbehenn and Martin 1992, Kopper et al. 2002). Barbehenn and Stannard (2004) demonstrated that the peritrophic envelope secreted by the midgut epithelium represents an important antioxidant that protects the midgut epithelium. Barbehenn et al. (2005) evaluated tussock moth response to phenolic compounds in sugar maple, *Acer saccharum* Marshall (Sapindales: Sapindaceae), and red oak, *Quercus rubra* L. (Fagales: Fagaceae), foliage. Results indicated that the complex mixtures of phenolics in leaves of these species have defensive oxidative activities that can damage larval midguts. However, oxidative activity was higher in sugar maple leaves than in red oak leaves, resulting in higher concentrations of semiquinone radicals in larval midgut fluids when fed on sugar maple than on red oak. Whitemarked tussock moth larval survival is not affected by ingested tannins (Kopper et al. 2002).

Although whitemarked tussock moth larvae appear to be tolerant of a variety of ingested plant defenses, Tallamy et al. (2010) reported that larvae were unable to survive on any of a suite of 20

alien plant species that have become naturalized and dominate forest understories in the mid-Atlantic states. These results suggested that other factors associated with insect–plant interactions over long time periods are important.

Variation in quality and quantity of food resources affects host preferences and larval growth and development rates. For example, [Kopper et al. \(2002\)](#) reported that larvae fed diets amended with condensed tannin showed prolonged development times, reduced relative growth rates, and reduced food conversion efficiencies. [Johns et al. \(2009\)](#) reported that young larvae feed exclusively on young foliage of balsam fir whereas older larvae feed on both young and mature foliage. They found that larvae had 32–65% higher fitness on diets composed of a mixture of foliage age classes, compared with fitness when fed a single age class of foliage. These results indicated that dietary mixing of different foliage age classes increased nutrient balance and/or diluted toxic defensive chemicals.

Environmental changes cause variation in foliage quality and quantity ([Mattson and Haack 1987](#), [Schowalter 2016](#)). [Broderon et al. \(2012\)](#) reported that larvae fed on hosts with higher nutritional quality showed lower mortality when infected with nucleopolyhedrosis virus (*Baculovirus*). Drought increased production of phenolic glycosides, but whitemarked tussock moth larval performance was not significantly affected ([Hale et al. 2005](#)). However, [Kopper et al. \(2001\)](#) and [Lindroth et al. \(2002\)](#) found that host preferences and larval performance could be altered by host exposure to carbon dioxide or ozone. [Agrell et al. \(2000\)](#) reported that elevated atmospheric CO₂ increased the concentration of defensive compounds (and reduced whitemarked tussock moth performance) in aspen, *Populus tremuloides* Michaux (Malpighiales: Salicaceae), an early successional species, under high-light conditions, and increased concentration of defensive compounds in paper birch, *Betula papyrifera* Marshall (Fagales: Betulaceae), and sugar maple, two late-successional species, under low-light conditions. These data indicated that rising atmospheric CO₂ concentrations could augment the ability of these tree species to defend against whitemarked tussock moth in their natural successional settings.

Mortality Factors

Whitemarked tussock moth larvae and pupae are preyed upon by a variety of invertebrate and vertebrate predators. In addition, larvae are parasitized by a variety of parasitoids and entomopathogens. [Howard \(1897\)](#) and [Hall and Buss \(2014\)](#) listed 17 species of tachinid (Diptera) parasitoids ([Fig. 6](#)), 26 species of hymenopteran parasitoids, and 11 species of predators that attacked whitemarked tussock moth larvae and pupae. They also noted that birds readily



Fig. 6. Tachinid puparium from tussock moth larva. Photo by Donald W. Hall, University of Florida, with permission.

fed on the larvae. [Medina and Barbosa \(2002\)](#) used open and closed cages to demonstrate that birds are the primary predators of large and small larvae. They noted disappearance of 60% of small larvae and 10% of large larvae in open cages. Small mammals typically are important predators of pupae ([Medina and Barbosa 2002](#)).

A variety of parasitoids attack whitemarked tussock moth larvae. [Medina et al. \(2005\)](#) compared parasitism by 10 parasitic Hymenoptera and Diptera species on tussock moth larvae feeding on willow, *Salix nigra* Marsh (Malpighiales: Salicaceae) and box elder, *Acer negundo* L. (Sapindales: Sapindaceae). Tussock moth larvae developed faster and reached larger pupal weights on willow, suggesting that parasitism rates on willow should be lower, due to shorter exposure time. Contrary to this hypothesis, total parasitism was higher on willow (70%) compared with box elder (30%). Only an unidentified *Casimaria* sp. (Hymenoptera: Ichneumonidae) showed higher parasitism rates on larvae fed box elder (86%) versus willow (14%).

Some, but not all, biological control agents released for management of gypsy moth also parasitize whitemarked tussock moth ([Raffa 1977](#), [Wallner and Grinberg 1984](#)). Whitemarked tussock moth is a suitable alternate host for the gypsy moth parasitoid *Apanteles porthetriæ* (Muesebeck) (Hymenoptera: Ichneumonoidea) ([Raffa 1977](#)) and *Rogas lymantriae* Watanabe (Hymenoptera: Braconidae) ([Wallner and Grinberg 1984](#)). [Fuester et al. \(2001\)](#) reported that a parasitoid, *Aphantorhaphopsis samarensis* (Villeneuve) (Diptera: Tachinidae), released for biological control of gypsy moth oviposited in whitemarked tussock moth larvae but did not successfully complete development.

[Guzo and Stoltz \(1985, 1987\)](#) reported that tussock moth hemocytes encapsulate and destroy eggs of *Hyposoter* spp. (Hymenoptera: Ichneumonidae). However, when larvae were previously parasitized by *Cotesia melanoscela* (Ratzeburg) (Hymenoptera: Braconidae), *Hyposoter* eggs were not encapsulated, and both parasitoids were able to complete development. Injection of tussock moth larvae with *Baculovirus*-like particles and venom from *C. melanoscela* produced the same results, demonstrating that obligatory multiparasitism is necessary for successful parasitism by some parasitoids.

Several entomopathogens parasitize whitemarked tussock moth larvae. [Lynn and Shapiro \(1997\)](#) and [Hall and Buss \(2014\)](#) reported that whitemarked tussock moth were susceptible to nuclear polyhedrosis virus (*Baculovirus*) and cytoplasmic polyhedrosis virus (*Cypovirus*). Larvae dying from *Baculovirus* infection characteristically hang limp by their prolegs ([Fig. 7](#)).

Whitemarked tussock moth larvae are vulnerable to application of *Bacillus thuringiensis* Berliner (Bacillales: Bacillaceae) var. *kurstaki* ([Rossmoore et al. 1970](#), [Morris 1973](#), [van Frankenhuyzen et al. 1992](#)). Among the *B. thuringiensis* endotoxins, larvae are vulnerable to Cry1Ab but show low sensitivity to Cry1Ac and Cry1D and no sensitivity to Cry1B or Cry1E ([van Frankenhuyzen et al. 1993](#)).

[Hajek et al. \(2000\)](#) reported that the fungus, *Entomophaga maimaiga* Humber, Shimazu and Soper (Entomophthorales: Entomophthoraceae), which infects gypsy moths and persists in soil around trees, also could infect whitemarked tussock moth larvae on understory vegetation and soil, especially when in prolonged contact with the soil. [Hajek et al. \(2004\)](#) subsequently reported that this entomopathogen infected whitemarked tussock moth only when gypsy moth populations were high and had high infection rates.

Outbreaks are likely controlled by predators, parasitoids and entomopathogens. [Wilson \(1991\)](#) reported that 88% of pupae were killed by predators and parasites in a black walnut plantation in Michigan during an outbreak in 1978–1981. [van Frankenhuyzen et al. \(2002\)](#) reported that infection rates by *Entomophaga aulicae*

(Reichardt) Humber (Entomophthorales: Entomophthoraceae) and a *Baculovirus* sp. reached 75% each during an outbreak in Nova Scotia during 1996–2001. Together the two pathogens accounted for 50% cohort mortality of tussock moth larvae.

Effects on Forest Ecosystems

Whitemarked tussock moths are common defoliators in eastern forests but are not generally considered to cause significant defoliation or harm large trees (Drooz 1985, Furniss and Carolin 1992, Hall and Buss 2014). Most trees are capable of replacing lost foliage after larvae have disappeared (Fig. 8). However, severe defoliation



Fig. 7. Tussock moth larva infected by nuclear polyhedrosis virus (*Baculovirus*). Photo by Donald W. Hall, University of Florida, with permission.



Fig. 8. New foliage production (compensatory growth) on southern live oak, *Quercus virginiana* Miller (Fagales: Fagaceae), in wake of defoliation. Photo by TDS.

and tree mortality can occur during outbreaks (Webster 1916, van Frankenhuyzen et al. 2002).

Webster (1916) documented an outbreak in Iowa in 1916. He noted that, whereas this tussock moth was common in the state, it rarely attracted such widespread attention. Elm, *Ulmus americana* L. (Rosales: Ulmaceae), “soft maples”, *Acer* spp. (Sapindales: Sapindaceae) and “linden” (basswood), *Tilia americana* L. (Malvales: Malvaceae), were most severely defoliated, although many other tree species, including apple, *Malus pumila* Miller (Rosales: Rosaceae), also were defoliated.

Outbreaks have occurred at about 15-yr intervals in the north-eastern United States and Canada since the 1960s and last 3–5 yr (Wilson 1991, van Frankenhuyzen et al. 2002). van Frankenhuyzen et al. (2002) reported that an outbreak between 1996 and 2001 covered a cumulative 2.4 million ha (5.9 million ac). At its peak, the infestation covered 203,000 ha (500,000 ac) in 1997 and 590,000 ha (1.5 million ac) in 1998. This species is capable of killing balsam fir after 1 yr of severe defoliation (van Frankenhuyzen et al. 2002).

Defoliation by whitemarked tussock moth may have little long-term effect on forests (Hall and Buss 2014). Hale et al. (2005) reported that herbivory by gypsy moth induced chemical resistance to further herbivory in black poplar, but no such resistance was induced by whitemarked tussock moth larvae. Wilson (1991) reported that natural enemies may have limited defoliation of individual black walnut trees to <5% during an outbreak in Michigan in 1978–1981.

Whitemarked tussock moth can affect carbon, nitrogen, and phosphorus fluxes in forests (Meehan and Lindroth 2007, 2009). Couture and Lindroth (2014) found that whitemarked tussock moth frass had higher concentrations of nitrogen and condensed tannins and lower C:N ratios than did litter. Frost and Hunter (2007) labeled whitemarked tussock moth frass with ^{15}N and demonstrated that red oak seedlings acquired the labeled N from frass during the same growing season, potentially offsetting any negative effects of defoliation. Frost and Hunter (2008) used ^{13}C and ^{15}N to show that herbivory by this species significantly reduced carbon allocation to fine roots by 63% and correspondingly increased C allocation to foliage. Herbivory also significantly reduced nitrogen allocation to fine roots by 39% but increased nitrogen storage in taproot and stem tissues. Hunter and Frost (2008) concluded that red oak seedlings responded to moderate herbivory by simultaneously increasing foliar C, maintaining C deposition to roots, maintaining N assimilation and shifting N resources to storage.

Frass deposition by this species alters litter decomposition rate and soil respiration. Hillstrom et al. (2010) manipulated whitemarked tussock moth frass in microcosms at the Free Air CO_2 Enrichment (FACE) site in Wisconsin and found that large amounts of frass deposition doubled the rate of soil carbon flux and nitrogen immobilization. However, the treatments with small amounts of frass had little effect on these fluxes.

Effects in Plantations and Blueberry Crops

Wilson (1991), Wagner (2005), Isaacs and van Timmeren (2009, 2012) and Plett et al. (2010) reported that whitemarked tussock moths can cause severe defoliation in walnut orchards, poplar and Christmas tree plantations, and blueberries during outbreaks. Young plants can be killed (Isaacs and van Timmeren 2009). In addition, pickers can suffer from severe tussockosis (see next section), and larvae collected during mechanical harvesting can cause significant economic losses for producers (Isaacs and van Timmeren 2009, 2012).

Effects on Public Health

Urticating hairs and setae connected to venom glands are found in tussocks and hair pencils (Gilmer 1925). The hairs on larvae or in cocoons frequently cause dermatitis and allergic reactions in sensitive individuals (Fig. 9), a condition known as tussockosis (Knight 1922; Gilmer 1925; Goldman et al. 1960; Diaz 2005; Wagner 2005; Hossler 2009, 2010; Hall and Buss 2014). Hairs in cocoons retain their urticating capability for a year or more (Hall and Buss 2014).

Management

Management Options in Forests and Plantations

Early detection is a key to effective management. Pheromone traps can be used for detection and monitoring for population growth. Isaacs and van Timmeren (2009) compared pheromone trap designs and concluded that large plastic delta traps (Scentry Biologicals Inc., Billings, MT) were the most effective for trapping adult male moths through the season. Pheromone lures remaining in traps all season maintained similar effectiveness to those changed regularly, suggesting no need to replace lures.

Grant (1978) demonstrated in field trials that the Douglas-fir tussock moth pheromone, (Z)-6-heneicosen-11-one, could disrupt mating in whitemarked tussock moth populations. Grant et al. (2006) subsequently reported that, while (Z)-6-heneicosen-11-one is a minor component of the whitemarked tussock moth pheromone, (Z,Z)-6,9-heneicosadien-11-one is a much more powerful sex pheromone. However, this compound is thermally unstable under field conditions, making baited traps effective for only one night. This problem was solved by using a stable pheromone precursor, (Z,Z)-6,9-heneicosadien-11-one ethylene ketal, which is hydrolyzed to the pheromone form by an acidic aqueous solution (2% *p*-toluenesulfonic acid in 35% aqueous sorbitol), and using a small, autonomous pump (Med-e-Cell Infudisk, Med-e-Cell, San Diego, CA) to deliver the precursor continuously to a suitable substrate where it is converted rapidly into the attractive pheromone component (Grant et al. 2006, 2008). Pheromone traps for monitoring whitemarked tussock moth populations are available from Great Lakes IPM, Inc. (Vestaburg, MI, <http://www.greatlakesipm.com/whitemarkedtussockmoth.html>) and Trécé, Incorporated (Adair, OK, <http://www.trece.com/index.html>).

In some cases, tussock moth abundance can be managed by manipulating host condition. For example, Plett et al. (2010) found that a transgenic poplar, *Populus tremula* L. x *Populus alba*

L. 717-1B4 (Malpighiales: Salicaceae), with genes for overexpression of trichome density showed greater resistance to whitemarked tussock moth larvae than did cultivars that did not express this trait. Plantations dominated by poplars expressing this trait should be less vulnerable to tussock moth outbreaks.

Options Available to Homeowners and Blueberry Growers

A number of management options are available to homeowners. As for prevention of other lepidopterous larvae, homeowners can reduce the vulnerability of host trees by maintaining good tree vigor, especially by keeping trees well-watered (Minnesota DNR 2017). Small trees can be protected by collecting and destroying egg masses (Webster 1916). Egg masses can be removed during winter when absence of leaves makes cocoons and egg masses most visible (Webster 1916).

Larvae and frass can be swept from walls and outdoor furniture with a stiff broom or pressurized water. Pupae may be more difficult to remove with pressurized water but can be removed with a stiff broom (Minnesota DNR 2017). Adhesive bands (such as Tanglefoot, Scotts MiracleGro, Marysville, OH) on tree trunks can entangle larvae crawling up or down trees (Webster 1916). Some products containing malathion and permethrin are labeled for application to lower foundations of homes to prevent larvae from crawling up walls (Meeker 2013, Minnesota DNR 2017, Krischik and Hahn 2018).

Insecticides

Noninsecticidal options are generally recommended over insecticides for this insect. When control measures are necessary, tussock moth larvae are generally controlled by applications recommended for control of other larvae, especially forest tent caterpillar, *Malacosoma disstria* Hübner (Lepidoptera: Lasiocampidae), and gypsy moth (with which whitemarked tussock moth commonly co-occurs) (Hall and Buss 2014, Isaacs and van Timmeren 2012, Louisiana Insect Pest Management Guide 2018). All insecticides should be used only according to their label directions and appropriate precautions taken to avoid exposure of humans, pets, livestock, or other nontarget organisms.

Control options vary in their environmental effects. Microbial insecticides, particularly *Bacillus thuringiensis* and *Baculovirus*, are effective and have minimal effects on nontarget species, although other species of Lepidoptera are vulnerable (Morris 1973, Ives et al. 1982, West et al. 1987, Meeker 2013). West et al. (1987) described successful reduction of whitemarked tussock moth defoliation in mixed stands of balsam fir and paper birch using a commercial formulation of *Baculovirus*. Diflubenzuron (an insect growth regulator) has minimal nontarget effects but should not be used near water, where it can affect aquatic arthropods adversely (Krischik and Hahn 2018). Other biorational insecticides include azadirachtin (a botanical insecticide) and insecticidal soap (Krischik and Hahn 2018). Conventional insecticides include several pyrethroids (such as bifenthrin and permethrin), organophosphates (such as acephate and malathion), and carbamates (carbaryl) (Krischik and Hahn 2018).

Obviously, control in blueberries is more complicated because any control options must be compatible with human consumption of the crop. Pheromone traps placed around the perimeter of blueberry fields adjacent to woodland in early June facilitates detection of moths flying into the blueberries (Isaacs and van Timmeren 2012). Isaacs and van Timmeren (2009) reported two generations of larvae in blueberries in southwestern Michigan. First (spring) generation larvae hatched during bloom and were



Fig. 9. Pruritic welts and erythema caused by urticating hairs from tussock moth. Photo by Donald W. Hall, University of Florida, with permission.

present on bushes from late May until early July, starting at growing degree-day (GDD) accumulation of 206 GDD above a threshold of 12.8°C. Egg hatch of the second (summer) generation started at 1157 GDD (above a threshold of 12.8°C), or 318 GDD after the first sustained capture of first generation male moths emerging in June–July. Early control is critical because as larvae become larger, they become more difficult to control. If larvae are detected near harvest, apply an appropriate and effective insecticide with short preharvest interval and short re-entry interval. Good coverage of the whole bush is necessary to ensure that the insecticide directly contacts all larvae (Isaacs and van Timmeren 2012). However, larvae are sensitive to a wide variety of insecticides so are likely to be controlled by applications made for other blueberry pests (Isaacs and van Timmeren 2012).

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