Integrated Pest Management Reviews **4:** 1–20, 1999.

Monitoring of pear psylla for pest management decisions and research

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Received 6 May 1998; accepted 3 November 1998

Abstract

Pear psylla, *Cacopsylla pyricola* (Foerster) (Homoptera: Psyllidae), is one of the key insect pests in North American pear production. In some growing areas, more than 50% of dollars spent to control arthropod pests in commercial pear are directed specifically at controlling this species. Control measures require accurate and timely information about dispersal, onset of egg-laying in spring, densities in the orchard, and age composition of the population. To meet these ends, a number of sampling methods have been developed to monitor pear psylla, the most common being (for pest management purposes) visual inspection of spurs and foliage for nymphs and eggs, and use of beat trays to monitor adults. Action thresholds have been developed for counts obtained with either method. However, threshold estimates are fairly narrowly defined, referring to a somewhat limited group of pear cultivars, type of injury to be prevented, and pest management program being used. Further refinement has been difficult because of an incomplete understanding of psylla's spatial distribution, seasonal changes in spatial distribution, and unknown or seasonally changing action thresholds. Beat trays and visual inspection of foliage have also been used to monitor pear psylla in various types of research projects, including studies of dispersal and biological control. Other sampling tools used in research include sticky traps, suction traps, and water pan traps. Density estimates obtained by these different sampling methods tend be positively correlated, albeit with high levels of unexplained variation. For counts obtained by sticky traps, much of the unexplained variation can be attributed to flight activity of the insect, which is known to depend (at a minimum) on sex, morphotype, reproductive status, time of year, time of day, leaf fall, and weather. Thus, if sticky traps are to be used in a pest management program for pear psylla, it must be recognized that counts on traps will include both density and (potentially large) behavioral components. I conclude this review with suggestions about the type of research that would improve monitoring techniques for this pest and assist eventually in developing a more effective control program.

Introduction

At least seven species of pear (*Pyrus*)-feeding psyllids in the genus *Cacopsylla* are recognized from the western Palearctic region (Burckhardt and Hodkinson 1986). Several of these species are important pests of commercial pear, most notably *Cacopsylla pyricola* (Foerster) in North America and Europe, and *C. pyri* (L.) in Europe. *Cacopsylla pyricola* probably originated in western Eurasia in contact with wild *Pyrus* (Hodkinson 1984, Burckhardt and Hodkinson 1986). The species was introduced into the eastern United States in the early 1800s apparently on infested pear seedlings shipped from Europe (Slingerland 1892), and rapidly dispersed from the east coast of the United States to the pear growing regions of the western U.S. The species arrived in the western United States in the early 1900s (Webster 1939, Anonymous 1946). At the present time, *C. pyricola* – with codling moth (*Cydia pomonella* (L.)) – is the most damaging insect pest of pears in the major pear growing regions of North America.

In this review, I summarize various topics associated with monitoring *C. pyricola* (hereafter referred to as pear psylla). This pest is extremely difficult to control, and effective management requires an investment in efficient and timely sampling for adults and immatures (Brunner 1982). Much of the recent literature and research concerning pear psylla focuses on populations occurring in the Pacific northwest region of North America, and this review necessarily emphasizes these populations and growing regions. Some reference will be made to populations occurring in central California, the eastern United States, and Europe, and I will also briefly discuss aspects of sampling for the congeneric pear psyllid, *C. pyri*, occurring in Europe. The review will first describe pear psylla life history, types of injury, and treatment thresholds. Pear psylla has a fairly complicated life history and causes several types of damage to the pear tree, and these characteristics complicate sampling, as I will show. Second, I will summarize practices currently used to control this pest, because methods used to control pear psylla partially affect decisions about techniques used to monitor the insect. For example, control programs that are directed at the overwintered adult require a different method of monitoring than control practices that specifically kill eggs. Third, I will describe the different types of tools used to monitor pear psylla in pest management programs and will provide sample sizes recommended to make management decisions. Fourth, I will contrast different sampling tools in terms of what actually is being monitored. This section will discuss whether different sampling methods lead to similar conclusions about psylla densities (i.e., how strongly correlated are different methods?). Some of this discussion will critique the use of sticky traps for monitoring pear psylla, and will propose that this method of sampling probably is more useful for monitoring flight activity and dispersal than for monitoring psylla density. Lastly, I will conclude with some comments about research areas that should be pursued.

Life history of pear psylla

This discussion of life history refers primarily to populations occurring in central Washington (latitude 46°N); however, aside from some variation in timing of certain events or in number of generations, the description applies fairly generally to pear psylla from other regions. The species is seasonally dimorphic (Wong and Madsen 1967, Oldfield 1970), producing a large dark overwintering adult (winterform) beginning in early September (Figure 1) that is quite distinct from the smaller and light-colored summerform adults. This is a true seasonal dimorphism, controlled by photoperiod (Wong and Madsen 1967, Oldfield 1970), and appears to be an uncommon trait in *Cacopsylla* (most *Cacopsylla* spp. are univoltine and hence lack seasonal dimorphism; see Jensen (1951), Hodkinson and White (1979), and Ossiannilsson (1992) for information about voltinism in *Cacopsylla*). Nymphs that experience short-days produce winterform adults, whereas those that experience long-days produce summerform adults. The dimorphism is striking enough that the two morphotypes were at one time considered to be distinct species (Slingerland 1892). The two morphotypes also differ physiologically and behaviorally, and these differences complicate monitoring efforts (see below).

Pear psylla spends much of the winter in reproductive diapause, characterized by immature ovaries and a lack of mating. The winterform adult overwinters both on the pear host plant and away from the host plant. Dispersal of winterforms from the orchard in autumn begins in mid- to late-September (Figure 1), and peaks during late-October and early-November (Horton et al. 1994b), coinciding with leaf fall in pear (approximate timing for major phenological events in pear is summarized in Table 1). A variety of non-host plant species have been shown to provide suitable overwintering habitats (Kaloostian 1970, Horton et al. 1994a). The proportion of an orchard's population that overwinters in the orchard rather than dispersing is not known and appears to vary among years even at the same orchard (Horton et al. 1994b). Cool, wet autumns result in a reduction in dispersal out of the orchard. Reentry into the pear orchard begins in February (Figure 1) well before the appearance of foliage in pear (Horton et al. 1992). Psylla disperse among orchards between the autumn and spring, thus individual orchards may exhibit a large change (either negative or positive) in peak density between fall and spring (Horton et al. 1992).

Egg-laying by overwintered winterforms begins in late February or early March (Figure 1). Because of the absence of foliage at this time (Table 1), the first eggs are deposited directly in wood, generally at the base of fruit and leaf buds. As foliage becomes available beginning in mid- to late-March (Table 1), oviposition shifts to occur primarily on expanding leaves and flowers. Fecundity of the overwintered adult appears to be quite high, at least as estimated by one laboratory study (mean fecundity = *ca*. 1000 eggs per female [maximum >2500 eggs]; Horton and Lewis 1996). Others (Georgala 1956, McMullen and Jong 1977) have provided lower estimates. High temperatures cause a substantial reduction

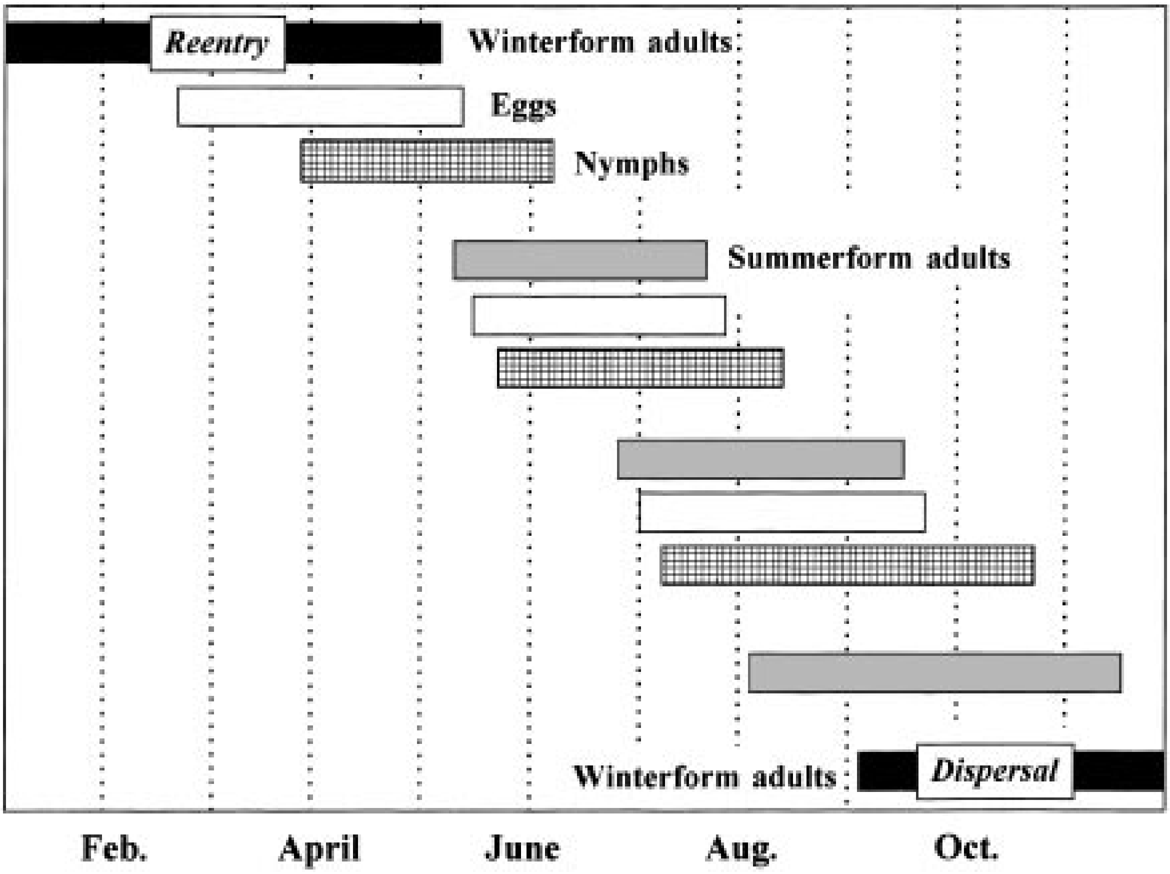

Figure 1. Approximate phenology of pear psylla in central Washington pear orchards.

in fecundity (McMullen and Jong 1977). Estimates of fecundity or egg-laying potential for winterforms of the congeneric psyllid *C. pyri* are available in Kapatos and Stratopoulou (1996) and Lyoussoufi et al. (1988).

Offspring of the overwintered generation eventually eclose as summerform adults, first appearing in mid-May (Figure 1). Pear psylla has 2–4 summerform generations per season in most pear growing regions. Summerform adults tend to oviposit on rapidly growing leaf tissues, often placing their eggs along the leaf mid-vein (Georgala 1956, Horton 1990). Fecundity of summerform adults varies (depending upon the study) between *ca*. 140 and 600 eggs per female (Georgala 1956, Burts and Fischer 1967, McMullen and Jong 1977). Field-based estimates of fecundity range between 50 and 180 eggs per female (McMullen and Jong 1972).

Spring- and summer-deposited eggs require *ca.* 6–10 days to hatch, depending upon temperature (McMullen and Jong 1977). There are 5 nymphal instars. Nymphs require 3–4 weeks to complete development at moderate (21–27°C) temperatures (Georgala 1956, McMullen and Jong 1977). Male and female progeny are produced in equivalent numbers (Burts and Fischer 1967). Feeding nymphs are immersed in pools of honeydew, which they produce in extremely large amounts. Older nymphs are fairly mobile and may move around on the leaf surface or petiole between feeding bouts. In central Washington, summerform adults that eclose in August and September produce offspring that eventually eclose as winterform adults (Figure 1). These winterforms enter reproductive diapause for overwintering. Some third generation summerform adults may be produced in certain years (Figure 1). Eggs and nymphs produced by these adults

Table 1. Approximate timing of major phenological events for 'Bartlett' pear trees in central Washington

Stage	Timing
Dormant	Winter to mid-March
Overwintering stage; no spread of bud scales	
Delayed dormant	Early- to mid-March
Bud scales beginning to spread; some green visible	
Clusterbud/pre-pink	Late-March to early-April
Flowers within a single cluster just beginning to separate	
Pink	Late-March to mid-April
Flowers within a single cluster fully separated; petals on flowers beginning to show	
Full bloom	Early- to mid-April
Petal fall	Mid-April to early-May
Harvest	August
Leaf fall	Beginning in late-October

probably would not complete development before leaf fall or the onset of winter temperatures. Eggs, nymphs, and summerform adults do not survive the winter in the central Washington study region.

Monitoring pear psylla is made difficult by the uneven distribution of insects on the trees, a distribution that may in fact change seasonally. The most noticeable seasonal shift in distribution is that by the egg-laying adults, discussed to some extent above. The first eggs in each season are deposited on wood at the base of unopened buds. Egg-laying then shifts in spring to opening buds, and then shifts again in summer to rapidly growing leaves occurring on terminals or water sprouts (Westigard et al. 1979, Fye 1982, Brunner 1984). Consequently, distribution and location of eggs and nymphs change seasonally (Westigard et al. 1979, Fye 1982). Densities of psylla may also vary with height in the tree canopy. During the summer, densities of eggs, nymphs, and adults are higher in the upper canopy than in the lower canopy (Brunner 1984, Horton 1994). Similar trends have been noted in early spring for *C. pyri* (Stratopoulou and Kapatos 1992). Densities of winterform adults in the fall are higher in the upper canopy than the lower canopy before leaf fall but not following leaf fall (Horton et al. 1993). Leaf fall prompts dispersal of psylla both within and between trees, resulting in a more uniform distribution of adults in the tree following leaf fall than before leaf fall (Horton et al. 1993).

Pest status

Pear production and control costs in the northwestern United States

In 1996, Washington, California, and Oregon ranked 1, 2, and 3, respectively, among the 50 states in acres of pear production (USDA-NASS 1997). Growers in these 3 states farmed 65,200 acres of pears, accounting for over 90% of the total U.S. acreage under pear production (USDA-NASS 1997). Almost 0.8 million tons of pears were harvested from Washington, Oregon, and California in 1996. Crop values in 1996 for these 3 states amounted to almost $300 million, representing 96% of the value for the total U.S. crop. Of these dollar returns, pears for canning or juicing (rather than for fresh market) amounted to *ca*. 15, 10, and 50% of the market returns for Washington, Oregon, and California, respectively (USDA-NASS 1997). These percentages are important to note, because economic thresholds for pest control depend substantially upon ultimate destination of the fruit (see below).

Pest control costs in northern Washington pear orchards have dropped from $850–1000 acre^{-1} in 1989 to $500–600 acre^{-1} at the present time, largely because of the availability of some new products to use against pear psylla (cost estimates obtained from discussions with local growers). The largest percentages of costs in many regions are spent to control pear psylla and mites, accounting for *ca*. 80% of total costs required to control pest arthropods in Oregon pear orchards (Westigard et al. 1986). In Medford, Oregon, more than 50% of total arthropod control costs are directed at pear psylla (R. Hilton, personal communication 1998). Currently, a standard program for pear psylla costs Washington and Oregon growers approximately $200–300 acre^{-1} (Hilton, personal communication 1998), although these costs vary among growers depending upon pest pressures, destination of fruit, and type of control program.

Types of damage

Pear psylla causes three primary types of damage: fruit russet, psylla shock, and pear decline (Burts 1970, Westigard et al. 1979, University of California 1991, Beers et al. 1993). Of these types of damage, russet is of most concern to growers, and control programs are generally directed at preventing this injury (Burts 1988). Grower emphasis on preventing russet injury is due to the fact that it can be caused by relatively low population densities (Burts 1988), unlike densities required to cause psylla shock, and because pear decline has rapidly diminished in importance in most pear growing regions.

Fruit russet is caused by the feeding activities of nymphs. As in other Homoptera, pear psylla ingests excessive quantities of plant juices and other plant products that must be eliminated (as honeydew) during the digestive process. Adult psylla excrete these waste products as small, waxy pellets that cause no harm to the plant or fruit. Conversely, immature pear psylla excrete large quantities of honeydew in the form of a syrupy liquid, and the product in this form is quite damaging. If nymphal-produced honeydew is in contact with fruit for a significant period of time it causes dark blotches or streaks on the surface of the fruit (russetting), which in turn results in downgrading of the fruit at harvest (Burts 1970). The damage may be exacerbated by a sooty mold fungus that colonizes honeydew and also marks fruit (Burts 1970); at high densities, the fungus may additionally cause a reduction in photosynthesis.

A second type of injury, also caused by nymphs, is of a more indirect nature than that previously mentioned. At high densities, pear psylla causes tree stunting, premature leaf drop, reduced fruit size, and premature fruit drop, resulting in substantial losses in yield (Burts 1970, Westigard and Zwick 1972). These symptoms have collectively been termed psylla shock, and are caused by a toxin in the saliva of feeding nymphs (Beers et al. 1993). Symptoms of the injury can be similar in appearance to those associated with pear decline disease. However, psylla shock is observed in pear growing regions not known to harbor the pear decline organism (Westigard and Zwick 1972), and, moreover, seems to be associated specifically with very high densities of nymphs. Psylla shock can be particularly damaging because the effects are not always restricted to the year of infestation. Symptoms may actually carry-over into a second year even if densities are not high the second year (Beers et al. 1993). Cultivars that are less preferred by psylla, such as some red pears or pears of Asian origin, tend not to experience high densities of pear psylla and thus may be less likely to experience this type of damage than more preferred cultivars.

Finally, adult pear psylla vector the mycoplasma-like organism (Hibino and Schneider 1970) that is responsible for pear decline disease (Lindner et al. 1961, Jensen et al. 1964). The pathogen causes sieve-tube necrosis at or below the graft union (Batjer and Schneider 1960, Westigard et al. 1979), preventing tree-synthesized nutrients from reaching the roots and resulting in starvation of the roots (Wilde and McIntosh 1964). Symptoms of the disease include a slow to abrupt decline or collapse in growth and vigor, causing a reduction in yield and (often) death of the tree. Pear trees that are affected by ‘slow decline’ may recover if psylla densities are kept low (Beers et al. 1993). The disease killed several million pear trees in the western U.S. during the 1950’s and 1960’s (Batiste and Bulla 1980). Severity of the disease depends upon psylla density and type of rootstock (Beers et al. 1993). Cultivars that have been grafted onto *Pyrus communis* rootstock are less susceptible than those grafted onto *P. pyrifolia* or *P. ussuriensis* rootstock. Resistant rootstock has largely remedied this problem in the western United States (Beers et al. 1993).

Factors affecting treatment thresholds

Decisions about whether control measures are required should be based upon the treatment or action threshold, i.e., that population density at which control measures are necessary to prevent eventual economic damage. Thus, before a monitoring plan can be implemented to provide control decisions, information about these thresholds is required. For pear psylla, sampling recommendations currently being used in management programs are often based as much upon experience and conversation with growers as upon systematic research. One reason for this shortcoming is the difficulty in estimating a treatment threshold for this pest.

As noted above, pear psylla causes three types of damage: russet, psylla shock, and pear decline. Because the pest causes multiple types of damage, there is no single treatment threshold for this species (Westigard and Zwick 1972). Depending upon the type of damage to be prevented, the threshold density may be comparatively high or low. For example, psylla shock is generally assumed to require quite high densities of nymphs (Westigard et al. 1979), whereas economic costs associated with russet begin to occur at nymphal densities that are comparatively fairly low (depending upon cultivar; see below). Life history stage of the insect is also important. Thus, russet is caused by nymphs but not by adults, whereas pear decline is spread between trees and orchards by the highly mobile adult stage. In sum, it is obvious that sampling plans and action thresholds designed to manage russet would differ from those designed to manage pear decline or psylla shock.

The situation is simplified somewhat because russet tends to be the injury of importance to growers. However, even for this single type of damage, thresholds are difficult to estimate. First, grower tolerance for russet depends in part upon pear cultivar. Naturally russetted cultivars such as 'Bosc' are less susceptible to early-season damage due to honeydew than are clear-skinned varieties such as 'Comice' and 'Anjou' (Westigard et al. 1981). For example, early season densities of nymphs necessary to cause 5% downgrading of fruit were estimated to be 2 and 0.4 nymphs per leaf for 'Bosc' and 'Anjou' cultivars, respectively (Westigard et al. 1981). Second, action thresholds for russet vary with destination of fruit. Pears that are to be canned or made into juice tolerate substantially higher levels of russet damage than pears that are to be sold as a fresh market product. Third, grower practices affect fruit susceptibility. For example, growers that use over-tree irrigation may suffer less russet than those using under-tree irrigation even at similar densities of pear psylla and for the same cultivar, due to the effects of water washing honeydew off of the fruit (Westigard et al. 1979). Brunner and Burts (1981) showed that tree washes, i.e., application of water or water and surfactant with a speed sprayer, significantly reduced russet in 'Bartlett' pear. Winfield et al. (1984) noted that wet weather in late summer reduced russet in pear that was growing in southeastern England.

Finally, age of the fruit when it is contacted by honeydew may affect extent of the damage, indicating that thresholds vary seasonally. It seems to be generally accepted that early-season injury is more costly than later-season damage, and that thresholds are thus lower early in the season (see Westigard et al. 1979). However, there are few quantitative data directly supporting this statement, and thus there is no strict consensus yet about the generality of this conclusion. For example, the natural russet of 'Bosc' fruit makes this cultivar less likely to be damaged by honeydew early in the season. However, late in the season, honeydew and sooty mold are very difficult to remove from the rough skin of the fruit, hence this cultivar is actually very susceptible to damage by russet late in the season (R. Hilton, personal communication 1998). For other cultivars, older fruit may be less susceptible to russet than young fruit because pear fruit develops a waxy cuticle as it grows, and this waxy cuticle may protect the fruit to some extent (Burts 1988). Also, early-season fruit generally projects upward, such that the fruit tends to be above any psylla nymphs that are feeding at that particular spur. As fruit ages and becomes heavier, it hangs below leaves and may more likely be damaged by falling honeydew (see full discussion in Burts (1988)). Data from Table 1 in Westigard and Zwick (1972) indicate that high nymphal densities early in the season caused more marking of 'Anjou' fruit than high nymphal densities near harvest, as discussed more fully below.

The quantitative relationships between density and yield loss to psylla shock or to pear decline are unknown. On susceptible rootstock, and with disease-carrying adult psylla, densities of pear psylla that cause economic damage due to pear decline are thought to be very low (Westigard et al. 1979). Injury due to psylla shock is generally thought to require high densities of nymphs, perhaps occurring over a period of several years (Westigard et al. 1979).

Control tactics

Commercial pear growers rely primarily on use of synthetic chemicals to control pear psylla. Unfortunately, these methods are not always entirely effective – especially during the summer generations – as pear psylla has become resistant to most classes of insecticides (Riedl et al. 1981, Follett et al. 1985, Burts et al. 1989, Croft et al. 1989). There are orchard practices that can be used by growers that make chemical controls more effective, primarily methods that cause the tree to be less suitable for growth and reproduction, or that cause the tree to be less attractive to ovipositing females. One common practice to reduce tree suitability is the use of only enough nitrogen fertilizer to satisfy requirements for good yield (Beers et al. 1993). Overuse of nitrogen causes excess tree vigor, and highly vigorous trees are very suitable for growth and reproduction of pear psylla (McMullen and Jong 1972, Pfeiffer and Burts 1983). Excess nitrogen has been shown to result in increased damage caused by psylla (Pfeiffer and Burts 1983). Similarly, trees should not be over-pruned, as this also may prompt excess growth of highly nutritious foliage (Westigard et al. 1979, Beers et al. 1993). Suckers or water sprouts should be removed from scaffold limbs (Beers et al. 1993), because these are a source of rapidly growing and highly nutritious foliage. Westigard et al. (1980) used a plant growth regulator to cause reduced growth of terminal shoots, and showed that the reduced growth was accompanied by lower psylla densities and a reduction in damage to fruit by honeydew. Lastly, the tree surface can be modified by using horticultural mineral oil to force delays in oviposition (see below), and this assists in chemical control of the immature stages.

Current control recommendations emphasize destruction of the overwintered generation or offspring of the overwintered generation with insecticides (Westigard and Zwick 1972, Beers et al. 1993). Successful control in late-winter and early-spring is necessary to prevent problems later in the growing season. A typical control program for overwintered adults is characterized by an application of horticultural mineral oil, sulfur, and insecticide (often a synthetic pyrethroid or chlorinated hydrocarbon) in late winter at the dormant stage in pear, repeated as necessary at the delayed dormant and clusterbud stages (Table 1) of pear (Westigard et al. 1979); the post-dormant sprays may or may not include the sulfur and oil treatments. An areawide approach involving synchronous spraying of orchards may improve prebloom control (Burts 1968). Objectives of these early season controls are to reduce densities of the overwintered adult as much as possible. With good coverage, the dormant and delayed-dormant oil treatments also cause delays in oviposition by survivors or post-spray immigrants (Zwick and Westigard 1978). This delay, in turn, results in synchronization of egg hatch in early spring, which makes subsequent chemical controls – if they are necessary – more effective (Beers et al. 1993).

Some growers have recently begun to eliminate insecticides from the dormant and post-dormant oil applications, substituting a delayed dormant or clusterbud application of the insect growth regulator Comply (fenoxycarb; Ciba-Geigy) (Hilton and Westigard 1994, Horton 1996). This material is an exceptionally good ovicide (McMullen 1990, Horton 1996, Horton and Broers 1997) but appears to have little direct toxicity against the adult. With good coverage and appropriate timing, fenoxycarb is an extremely effective chemical to use against prebloom psylla populations (Hilton and Westigard 1994). One largely unexplored consequence of this shift in control methods is the effect it should have on the sampling program. Growers that use fenoxycarb as a substitute for the more traditional program may find that densities of adults and eggs are uncomfortably high. However, with good spray coverage and timing, the eggs will not hatch (McMullen 1990, Horton 1996, Horton and Broers 1997). Sampling methods that monitor adults or eggs could easily lead to overestimates of damage potential, unless it is recognized that the target of the spray is the unhatched egg.

With either management approach, correct timing of the dormant oil or oil + insecticide spray is critical for control (Hamilton 1948, Westigard and Zwick 1972). The first spray application should be delayed until reentry is mostly complete, but should not be delayed much beyond the onset of egg-laying (Hamilton 1948). A long-term sampling program in central Washington indicated that growers generally time their first spray only slightly earlier than complete reentry (Horton et al. 1992), coinciding about with the onset of egg-laying. There have been modest attempts to develop degree-day models that predict onset of egg-laying (Westigard and Zwick 1972, Brunner 1984) and timing of reentry (Horton et al. 1992), with aims toward improving timing of the dormant spray. Currently, however, sampling of the adult population is necessary to determine the onset of reentry in late winter, and sampling of fruit spurs for eggs is often the easiest way to determine the beginning of egg-laying. Methods used for these purposes are described below. Dissections of females collected in January and February may also be useful for predicting the onset of egg-laying (Westigard and Zwick 1972, Beers et al. 1993). The first eggs can be expected in the field when 15–20% of dissected females have mature eggs in their oviducts (Westigard and Zwick 1972).

Summer management of pear psylla is very difficult if the overwintered population is not controlled. Pear psylla is now resistant to many insecticides (Croft et al. 1989, Pree et al. 1990). Summer chemicals that are used include pyrethroids and newer products having novel chemistries (e.g., Agrimek [avermectin; Merck]: Burts 1985, Washington State University 1998). Sprays are most effective if directed against young nymphs (Beers et al. 1993), and this requires constant monitoring of the population. Brunner (1984) presents degree-day estimates predicting the initial appearance of first generation nymphs in north-central Washington, and this method may be useful for timing some control measures. Phenology of *C. pyri* based upon degree-day accumulations has been presented by Beránková and Kocourek (1994).

Monitoring pear psylla for pest management decisions

A variety of methods have been used to sample pear psylla, although the most commonly used tools for management programs have been beat trays ('limb jarring') to monitor adults, and visual inspection of spurs and foliage to monitor immatures. In this section, I will describe both of these methods in some detail, and will provide sample sizes currently recommended for use in psylla management programs. At the end of the section, I will provide information about use, design, and construction of sticky traps to monitor pear psylla. Sticky traps are used only infrequently in psylla management programs, but have been quite common in some research studies with pear psylla.

Beat trays

The standard method for monitoring adult pear psylla is by jarring them from limbs onto a white, cloth-covered tray (Burts and Retan 1973). This monitoring technique is especially useful during the prebloom period to determine whether retreatment is necessary following an insecticide application directed at overwintered adults (Burts and Brunner 1981). The technique is also highly useful for monitoring spring reentry and emergence from pear overwintering sites by overwintered adults, information that helps in timing the dormant spray application. Also, beat trays are effective for monitoring natural enemies of pear psylla, particularly predatory Heteroptera, Neuroptera, Coccinellidae, and spiders (Madsen et al. 1963, Gut et al. 1982, Burts 1983, Westigard et al. 1986). Finally, beat trays have also shown to be useful in research projects, as I will discuss in the next section.

Tray size varies to some extent among workers, but the most commonly used size appears to be 45×45 cm (Burts and Retan 1973). The tray is covered with a white cloth, both to make the insect visible for counting and to act as substrate to which adult psylla cling while they are being counted (Burts and Retan 1973). The frame may be constructed of wood or aluminum (Burts and Retan 1973). Samples are taken by holding the tray beneath a horizontal limb that is 2–3 cm in diameter and that has 'an average complement of branches and spurs' (Beers et al. 1993). The limb is then sharply rapped 2–3 times with a length of stiff rubber hose; sections of old spray hose work well for this purpose (Burts and Retan 1973). Dislodged psylla cling to the cloth and are counted. Samples should be taken in the morning when temperatures are cool, as this minimizes movement on the tray by the insects and also reduces tendencies for psylla to fly away before they can be counted. Late-season monitoring of psylla can be somewhat difficult using beat trays because jarring the limbs may dislodge fruit or leaves (Adams and Los 1989, Horton et al. 1993). Also, it should be noted that we have no good quantitative information about the relationship between tray counts taken at different times of the year. For example, there is no reason to assume that a tray count of a certain magnitude taken in February, before the appearance of foliage, in any way predicts the same absolute density of psylla as the same count obtained in mid-summer when trees are fully leaved. This lack of information can be troublesome, particularly for some research questions (e.g., Horton et al. 1992).

Burts and Brunner (1981) presented dispersion statistics for adult pear psylla obtained for beating tray samples taken before foliage was on the tree (March–mid-April) and after the foliage had appeared (May–June). Samples indicated that psylla had a statistically clumped distribution, as estimated using 'mean-crowding' regression analysis; the slope coefficient for the regression was $\beta = 1.3$. There was some indication that early-season tray counts were more variable than those taken in May and June (Burts and Brunner 1981), perhaps because presence or absence of foliage affects spatial distribution of adult pear psylla (this has yet to be addressed systematically). Spatial distribution of adults estimated by beat trays was similar before pesticide treatment and following treatment (Burts and Brunner 1981).

In Washington and Oregon pear orchards, a recommended sample size for an average sized pear block (say, 10–20 acres) is 25 trays per block (Burts and Retan 1973, Beers et al. 1993). The samples should be taken at random locations throughout the block and should also be well-dispersed over the sampling area (Burts and Retan 1973, Burts and Brunner 1981). Beers et al. (1993) suggested that it may be useful to concentrate samples to some extent in areas of the orchard known to have a history of infestation, although care should be taken that this approach does not lead to unnecessary treatment (R. Hilton, personal communication 1998). Small blocks of pear may need fewer samples, whereas blocks larger than 25–30 acres may require more than 25 samples to ensure adequate coverage of the orchard.

Burts and Brunner (1981) developed a sequential sampling plan for early-season (March–June) adults that is useful for making decisions about whether retreatment is necessary to control overwintered adults. Stop lines were calculated for several error rates and a retreatment threshold of 0.2 psylla per tray.

Crude treatment thresholds have been proposed for beating tray samples (Table 2). These thresholds have not been developed through systematic study, but rather are based on field-experience and discussion with growers and fieldmen (Westigard et al. 1979, Burts and Brunner 1981). The few quantitative data available relating damage and psylla numbers indicate merely that tray counts and marking of fruit are positively correlated ($r = 0.76$; Adams and Los 1989). Estimates in Table 2 assume that the type of damage to be prevented is marking of 'Bartlett' fruit by honeydew (Westigard et al. 1979). Furthermore, it should be reemphasized that recommendations in Table 2 for prebloom populations of pear psylla (i.e., before mid-April) are based upon control strategies that kill adults, and may not be as useful for products such as fenoxycarb whose primary effect is in preventing hatch of eggs. Action thresholds for adults in a program that centers around the use of fenoxycarb during the prebloom period may be higher than those that are summarized in Table 2, although this remains to be determined.

Treatment thresholds are thought to be somewhat lower early in the season than they are later in the year (Table 2), apparently because it is assumed that early-season fruit are more susceptible to russet than later-season fruit. Westigard et al. (1979) suggested that a tray count of winterforms higher than *ca.* 0.1 per tray (2–3 per 25 tray sample) during the prebloom period justifies action. Beers et al. (1993) indicated that treatment should be followed immediately with another tray sample, and that a post-treatment count higher than 0.2 per tray justifies retreatment (Table 2). Using the sequential sampling plan developed by Burts and Brunner (1981), a cumulative total of 6–8 psylla in 25 trays indicates that the threshold for retreatment has been exceeded, assuming an error level of $P = 0.2$. After petal fall (i.e., during the summerform generations) thresholds are thought to be higher, ranging between *ca.* 1–2 per tray for 'Bartlett' pear in the northwestern United States (Table 2). In Connecticut 'Bartlett' pear orchards, Adams and Los (1989) suggest an action threshold of 1.0–1.2 psylla per tray during the summer months.

Recommended sampling protocols and thresholds are somewhat different for central California orchards. Extension personnel suggest that growers take 25 tray samples in early December, just after leaf fall, to determine what subsequent control measures are needed (University of California 1991). If counts are very low (0 or 1 psylla per 25 trays), a single dormant application of oil may be sufficient for prebloom control. Higher densities may require supplemental oil sprays or the addition of insecticide to the initial dormant spray, particularly if it is anticipated that dispersing psylla may be entering the orchard during the prebloom period (University of California 1991). Advisers also recommend sampling immediately after the first dormant spray. If tray counts fall between 1 to 4 psylla

Table 2. Approximate treatment thresholds for Washington and Oregon orchards to prevent russet of 'Bartlett' fruit. Modified from Table 37 in Westigard et al. (1979), unless otherwise noted

Treatment period	Approximate treatment thresholds
Adults (tray samples)	
Before dormant spray	1 per 10 trays or 2–3 per 25 trays
Retreatment decisions; prebloom period (Burts and Brunner 1981, Beers et al. 1993)	5 per 25 trays (6–9 per 25 trays based upon sequential sampling plan in Burts and Brunner 1981)
Delayed dormant to mid-May	1 per tray
Mid-May to mid-June	1 per tray
Mid-June to mid-July	2 per tray
Eggs and nymphs (spurs and leaves)	
Delayed dormant to clusterbud	1 egg per spur
Clusterbud to mid-May	1–2 eggs + nymphs per fruit cluster
Mid-May to mid-June	0.1 eggs + nymphs per leaf
Mid-June to mid-July	0.2 eggs + nymphs per leaf
Spring and summer; 'Bartlett' and 'Anjou' cultivars (Burts 1988)	0.3 nymphs per leaf

per 100 trays, a second oil application should be put on. If more than 4 psylla are counted in 100 trays, an insecticide should be added to the oil. Summer thresholds depend upon predator densities. If tray samples indicate that lacewings (Neuroptera) or anthocorid bugs are present, summer controls for psylla may not be necessary (University of California 1991).

Spur and leaf samples

Densities of eggs and nymphs are determined by examination of spurs and leaves. Again, for management purposes, sampling must be adjusted seasonally to provide useful information. Oviposition preferences of pear psylla change over the course of the growing season, and this forces a change in sampling units. Before much bud swell has occurred, most egg-laying occurs at the base of the unopened spurs, and these spurs should be the focus of sampling (Westigard et al. 1979). Samples taken this early in the season may be useful to help determine the onset of egg-laying by overwintered adults and thus help with timing the first spray application. Between the late clusterbud stage (Table 1) and initial hatch of second generation eggs (i.e., the interval between mid- or late-April and late-May in central Washington), counts are made by looking at leaves and fruit associated with spurs (Burts and Retan 1973, Westigard et al. 1979). Summer counts are made by looking at leaves associated with growing terminals and fruiting clusters.

Sample size recommendations for prebloom populations of eggs and nymphs are available from several sources. Recommendations are based more upon experience than upon research, primarily because there are no published data describing spatial distributions of eggs and nymphs of *C. pyricola* [Burts 1988; nymphs and eggs of *C. pyri* are highly clumped during the prebloom period (Deronzier and Atger 1980)]. For prebloom samples taken in Washington orchards, Burts and Retan (1973) and Beers et al. (1993) suggest that a minimum of 10 spurs should be collected from typical sized (10–20 acres) pear blocks. For large blocks (20+ acres), the sample size should be increased. Sampling should be concentrated in portions of the orchard with a history of infestation (Beers et al. 1993). At very low densities, nymphs may be concentrated in but a few sites on the pear tree (Burts 1988), and sample sizes larger than 10 spurs are necessary. Spurs that are to be sampled should be pointing horizontally or upward, but not downward (Burts and Retan 1973). In California pear orchards, 25 spurs or fruiting clusters should be examined on each sampling date beginning at the late delayed-dormant period (University of California 1991). If no psylla are found in that first sample of 25 spurs, sampling should be continued in increments of 25 spurs until 100 samples have been examined.

For later generations (June–September), leaf samples are taken (Burts and Retan 1973, Westigard et al. 1979, Beers et al. 1993). In Washington and Oregon pear orchards, a minimum of 50 leaves should be collected for an average sized block, chosen to contain a mix of spur and growing terminals (Burts and Retan 1973). Leaves collected from growing terminals should be taken from both the tip and base of the terminal. The leaf petioles should be included in the sample, as late instar nymphs often feed at the juncture of the petiole and the shoot. In California orchards, samples collected in May and June should consist of 20 shoots per block taken weekly (University of California 1991).

Spur and leaf samples are inspected with a hand-lens or taken to the laboratory to be examined under a dissecting microscope. Spurs should be carefully pulled apart, and each leaf, stem, and developing fruit examined for the presence of eggs and nymphs. If eggs are to be counted, the leaf midvein, in particular, should be examined. A mite-brushing machine may be useful for dislodging nymphs and eggs from leaves onto glass plates where they can be counted fairly easily (Burts 1988, R. Hilton, personal communication 1998). Nymphs may also wash off of leaves by dipping samples into 70% alcohol, as shown for other psyllid species (Elder and Mayer 1990). Large samples of foliage may be more efficiently processed using a Berlese-Tullgren funnel (Harris 1971). Harris (1971) showed that recovery of pear psylla nymphs with this system approached 100% if a low wattage (40 W) bulb was used. Recovery of nymphs was poor if 60, 75, or 100 W bulbs were used, presumably because nymphs desiccated before escaping to the collecting funnel (Harris 1971). This method may also be useful for extracting predators of pear psylla from spur or foliage samples (Wiltshire and Glen 1984).

Some quantitative data are available describing the relationship between nymphal densities and damage due to russet (Table 3), and these data have been used to provide action thresholds. Savinelli and Tetrault (1984) showed that season-long densities of nymphs (mean numbers per leaf; samples taken weekly between late March and early October) were highly correlated

Table 3. Regression equations relating nymphal densities to damage of fruit caused by russet. Savinelli and Tetrault (1984): regression from raw data provided in reference; Westigard et al. (1981): regressions estimated from data plotted in their Figure 1; Westigard and Zwick (1972): regressions from data provided in their Table 1; Burts (1988): regressions provided in publication

Regression equation	$P : \beta_1 = 0$	r^2	N	Reference
Bartlett: % fruit marked = $-2.0 + 7.7$(nymphs per leaf)[a]	0.001	0.85	10	Savinelli and Tetrault (1984)
Bosc: % fruit marked = $-1.2 + 24.5$(nymphs per expanded leaf)[b]	0.02	0.96	4	Westigard et al. (1981)
Anjou: % fruit marked = $1.9 + 40.5$(nymphs per expanded leaf)[b]	0.06	0.88	4	Westigard et al. (1981)
Bosc: % fruit downgraded = $-0.08 + 1.8$(nymphs per expanded leaf)[b]	0.048	0.91	4	Westigard et al. (1981)
Anjou: % fruit downgraded = $-1.2 + 17.8$(nymphs per expanded leaf)[b]	0.03	0.94	4	Westigard et al. (1981)
Anjou: % fruit marked = $4.3 + 0.40$(nymphs per 12 spurs in May)	0.003	0.85	7	Westigard and Zwick (1972)
Anjou: % fruit marked = $3.9 + 0.98$(nymphs per 12 spurs in June)	0.004	0.84	7	Westigard and Zwick (1972)
Anjou: % fruit marked = $4.1 + 0.34$(nymphs per 12 spurs in July)	0.002	0.95	7	Westigard and Zwick (1972)
Anjou: % fruit marked = $2.9 + 0.34$(nymphs per 12 spurs in Aug.)	0.30	0.20	7	Westigard and Zwick (1972)
Anjou: % fruit marked = $7.1 + 0.11$ (nymphs per 12 spurs in Sept.)	0.55	0.07	7	Westigard and Zwick (1972)
Bartlett: DI[c] = $-3.5 + 12.6$(nymphs per leaf)[d]	—	0.43	47	Burts (1988)
Anjou: DI[c] = $-3.0 + 10.3$(nymphs per leaf)[d]	—	0.85	21	Burts (1988)

[a] Season-long average nymphs per leaf.
[b] Biweekly samples from fruiting clusters; April–June.
[c] DI = damage index (see text).
[d] Peak nymphal density; season-long.

with percentage of 'Bartlett' fruit discolored at harvest ($r = 0.92$). A linear regression fitted to their data predicts that each density increase of 1 nymph per leaf results in an increase in damaged fruit of *ca.* 8 percentage points (Table 3). Regression equations fitted to data estimated visually from plots in Westigard et al. (1981) dramatically illustrate the importance of cultivar in susceptibility to russet (Table 3). For example, early-season nymphal densities of 1 per leaf caused an estimated 2% and 17% downgrading (U.S. # 2's + culls) of fruit for 'Bosc' and 'Anjou' cultivars, respectively. Early season densities necessary to cause marking on 5% of fruit is estimated to be 0.25 and 0.08 nymphs per leaf for 'Bosc' and 'Anjou' cultivars, respectively (Table 3). Westigard and Zwick (1972) presented data from 7 blocks of 'Anjou' pear showing the relationship between mean nymphal densities and fruit damage for density estimates made separately for each month between May and September (Table 3). Regressions fitted to these data indicate that nymphal counts (numbers per 12 spurs) in May, June, or July predicted fruit discoloration at harvest, whereas counts obtained in August and September failed to correlate with damage. These results reinforce the idea that early-season damage is more important than late season damage, and that thresholds should be adjusted seasonally.

Finally, Burts (1988) used regression analysis to determine the relationship between damage to 'Bartlett' or 'Anjou' pear and peak nymphal densities (Table 3). Samples were taken at biweekly intervals over the course of the growing season, with the first sample taken in May. Peak nymphal density (numbers per leaf) during the growing season was used as the independent variable in the regressions. A damage index that expresses injury as % of fruit downgraded was used for the response variable (with culls receiving double weight):

$$\text{DI} = (\text{\% fruit downgraded to U.S. \#2} + [2][\text{\%cull fruit}]).$$

Results suggested that the relationship between peak nymphal density and damage was similar between ‘Bartlett’ pear and ‘Anjou’ pear (Table 3). Burts (1988) concluded that a peak nymphal density of 0.3 per leaf produced detectable fruit damage, and suggested that action thresholds should be based upon this density. A second estimate of density was obtained by calculating accumulated nymphal feeding days (numbers per leaf, season-long). Regression lines fitted to these data indicated that accumulated nymphal feeding days of 18 or 9 days per leaf caused fruit damage to ‘Bartlett’ and ‘Anjou’ pear, respectively (Burts, 1988).

Specific sampling recommendations for use of spur and leaf samples in a psylla control program have been made. As noted above, the dormant spray should be timed to coincide with the onset of egg-laying, and spur samples may be useful for determining initial egg-laying in the field. For post-dormant intervals, quantitative estimates of action thresholds are available (Table 2). These estimates of thresholds are based upon a combination of research (Table 3) and discussion with fieldmen and growers. The thresholds once again assume that the injury to be prevented is marking of fruit by honeydew. For Washington and Oregon pear orchards, Westigard et al. (1979; see also Table 2) suggested that thresholds are somewhat lower early in summer (0.1 eggs + nymphs per leaf) than later in the summer (0.2 eggs + nymphs per leaf). The threshold of 0.3 nymphs per leaf suggested by Burts (1988) is based upon his regression analyses, as summarized here in Table 3. Burts (1988) noted that the threshold estimate refers to the minimum density causing damage and does not take into account other factors that should be considered in making treatment decisions (e.g., costs of treatment; potential dollar return).

In California, pest managers recommend treatment with oil if 1–5% of prebloom spur samples are infested with eggs or nymphs, and recommend treatment with oil plus insecticide if >5% of spurs are infested (University of California 1991). For shoot samples taken in May and June in cooler growing regions, recommendations are that oil sprays should be directed at early instar nymphs if 20% of 20 shoots are infested; in warmer areas, the threshold is reduced to 1 infested shoot per 20 samples. Mid-summer recommendations are based upon whether natural enemies are present. If shoots contain lacewing eggs or larvae, treatment may not be necessary even if psylla nymphs are present (University of California 1991).

Sticky traps

A third method of monitoring adult pear psylla is with use of sticky traps. This monitoring technique is probably of more use in research than as a part of a management program, although some information for the latter is also available. Trap size, material, color, and adhesive are highly variable among different studies, so only a few general recommendations can be made about using this monitoring tool. Furthermore, as discussed in the following sections, behavior of pear psylla may complicate how one should interpret counts on sticky traps.

Horton and coworkers (Krysan and Horton 1991, Horton 1993, 1994, Horton et al. 1993, 1994b, Horton and Lewis 1997) constructed traps out of 30.5 cm square sheets of clear plexiglass (0.3 cm thick), which were either painted yellow or left clear. The surface of each trap was made sticky by coating it with a thin layer of STP Oil Treatment. Traps were then hung from limbs 1.5 to 2.5 m above ground, and replaced with new traps at weekly intervals. Psylla were counted by visual inspection of traps. In some studies, insects were also categorized as to morphotype or sex (Horton and Lewis, 1997). The oil adhesive was very effective in trapping psylla for these studies, but it may be less effective in areas that experience more rainfall because oil may wash off of traps (Adams and Los 1989). I found that the oil was considerably easier to apply to traps than some of the more common adhesives such as Tangletrap. Large numbers of traps could be coated with oil in a short period of time using large paint brushes or paint rollers.

A number of other trap designs have been used. Purcell and Suslow (1984) constructed traps out of 13 × 26 cm rectangles of ‘bright yellow plastic sheeting’ (0.8 mm thick) that had been coated with Stickem Special. Several other workers have used plywood or posterboard cut into squares or rectangles (Kaloostian and Yeomans 1944, Wilde 1962, Fye 1983, Adams et al. 1983, Adams and Los 1989), generally painted either yellow or white. Trap size varies substantially among studies (between 12 × 23 cm and 30 cm × 2.5 m). Hodgson and Mustafa (1984) made traps out of plastic cylinders (15 cm diameter × 30 cm long), painted yellow and covered with acetate. The acetate was coated with ‘banding grease’ to make it sticky. Cylinders were mounted on poles, and the poles were placed in the orchards.

Adams et al. (1983) compared a number of fluorescent paints for effectiveness in monitoring summer generation pear psylla, and found that hues having reflectance peaks that mimic pear foliage were most attractive. That is, yellow or green hues were more attractive than blue, red, or black colors. Krysan and Horton (1991) showed that the foliage-like colors were attractive to pear psylla during the time of year that foliage was on the tree, but were not attractive in late winter when foliage was absent and winterform psylla were recolonizing the orchard. Wilde (1962) showed that psylla counts were larger on yellow traps than on white traps.

Sticky traps have been used only infrequently in management programs for pear psylla. One of the earliest uses of yellow traps for monitoring pear psylla occurred in the northwestern United States during the period that pear psylla was moving into the area from the east. Entomologists used yellow sticky traps to replace visual inspection of trees as a means for locating orchards that had been newly colonized by pear psylla (Davis 1943, Anonymous 1946). This trapping method allowed scientists to monitor large regions for the initial appearance of the pest, and provided some indication of dispersal rates in the region. Some research has also been done relating sticky trap counts to damage potential. Counts of spring and summer generation adults on yellow traps have been shown to be correlated with presence of eggs and nymphs on spurs, and positively correlated with the presence of sooty-mold russet on pear fruit (Adams et al. 1983). Adams and Los (1989) suggested that a yellow trap (15×20 cm sized traps) count of 4–7 summer generation adults per trap per day in 'Bartlett' pear warrants treatment to prevent unacceptable densities of nymphs in the following weeks.

Monitoring techniques for research studies

A variety of monitoring tools, including those discussed above, have been used in both basic and applied research projects to study pear psylla. Beat trays have been used to monitor autumn dispersal into non-pear habitats, providing information on phenology of dispersal and numbers leaving the pear orchard (Purcell and Suslow 1984, Horton et al. 1994b). Horton et al. (1992) used beat trays to compare densities of winterform psylla in the orchard in fall (pre-dispersal) and spring (post-reentry), and used these data to make inferences about overwintering loss and dispersal of insects among different orchards. Using similar methods, Horton et al. (1993) demonstrated that autumn populations of winterform pear psylla dispersed between pear cultivars within an orchard, apparently in response to patterns in leaf fall in the different cultivars. Beat trays are commonly used to determine efficacy in field-based insecticide trials (e.g., Westigard 1974, Hilton and Westigard 1994). Trays are also a convenient tool to use in monitoring both pear psylla and natural enemies in IPM or biological control studies (Madsen et al. 1963, Fauvel and Atger 1981, Gut et al. 1982, Burts 1983). Finally, trays in combination with aspirators are commonly used to collect pear psylla and their natural enemies for laboratory use (e.g., Krysan and Higbee 1990, Horton et al. 1994a, Horton et al. 1998).

Spur and leaf samples are also useful for addressing specific research questions, both basic and applied. Pesticide trials commonly require foliage samples to determine efficacy of different chemicals or the most appropriate timing to apply chemicals (e.g., Westigard 1974, Solomon and Fitzgerald 1987, Hilton and Westigard 1994). Effectiveness of biological control agents or different IPM programs for psylla control has been studied by sampling foliage for eggs and nymphs of pear psylla (e.g., Madsen et al. 1963, Gut et al. 1982, Burts 1983, Solomon et al. 1989, Horton et al. 1997). Stratopoulou and Kapatos (1995) estimated fecundity in the field for *C. pyri* by sampling spurs and leaves for eggs, and then combining these estimates with absolute counts of adults obtained with other sampling methods. Horton and Broers (1997) used spur samples to determine effects of fenoxycarb on egg hatch under field conditions.

Several studies have used sticky traps to monitor dispersal or flight activity of pear psylla. Horton et al. (1994b) made use of yellow sticky traps to monitor autumn dispersal into apple orchards by winterform psylla, and showed that dispersal rates peaked during intervals of warm, sunny weather combined with leaf fall in pear. Purcell and Suslow (1984) used yellow traps to study movement by psylla into peach orchards, and with these data suggested that pear psylla helps spread peach yellow leaf roll disease in peach. Others have used sticky traps to quantify or model dispersal as a function of distance from the source pear orchard (Westigard and Madsen 1963, Purcell and Suslow 1984, Hodgson and Mustafa 1984, Westigard and Hilton 1990, Horton et al. 1995). Not surprisingly, traps placed close to source pear orchards captured the largest number of psylla, whereas those at longer distances from the orchard captured fewer psylla. Horton et al. (1995) showed that a reciprocal model of the form:

$$\text{trap catch} = \beta_0 + \beta_1(1/\text{trap distance}),$$

where β_0 and β_1 are intercept and slope coefficients, respectively, provided a good fit to data collected for winterforms dispersing through non-orchard habitats. Horton (1994) used sticky traps to demonstrate that summerform psylla exhibited more flight activity in the upper tree canopy than in the lower canopy. Finally, yellow sticky traps have been used to determine diurnal trends in flight activity of pear psylla, with data showing that flight of summerform psylla peaked in the morning, whereas that of winterform psylla peaked at mid-day (Horton 1993).

Methods other than those previously described have been developed to monitor adult pear psylla with objectives being to address specific research questions. Suction traps have been used to monitor dispersal of summerform and winterform pear psylla over non-orchard habitats (Hodgson and Mustafa 1984). Horton et al. (1995) monitored dispersal using large, clear interception traps (0.9 × 1.8 m in size, lightly coated with an oil adhesive) that were placed at various distances surrounding a source pear orchard. Yellow water pan traps have been employed to monitor flight activity in the orchard (Hodgson and Mustafa 1984). Horton et al. (1993, 1994b) used clear water pan traps to show that winterform pear psylla were dislodged from the pear tree during leaf fall in pear. Dispersal out of the orchard peaked simultaneously with peak water pan catch. Finally, Bronnimann (1964), Stratopoulou and Kapatos (1995), and Horton (1994) used large mesh bags to estimate absolute densities (numbers per leaf) of adult pear psylla. For this purpose, the bags were carefully drawn over large branches of pear and then tied close at the open end. The bagged branches were cut down from the tree and taken to the laboratory where both psylla and leaves were counted. Horton (1994) used these data in regression analyses to determine whether beating trays or sticky traps were equally effective in predicting absolute densities of summerform psylla; beat trays were more effective at predicting absolute density than were sticky traps. Horton (1994) and Horton et al. (1993) also used this sampling method to quantify adult movement between heights in the tree canopy. Bronnimann (1964) employed this technique to monitor absolute densities of natural enemies in pear orchards, whereas Stratopoulou and Kapatos (1995) used estimates of absolute densities to calculate oviposition rates of *C. pyri* under field conditions.

Comparisons of sampling methods

Are counts obtained by different methods correlated?

Studies with pear psylla show that density estimates obtained by different sampling methods tend to be positively correlated, albeit with large amounts of unexplained variation. Potential sources of this unexplained variation are discussed in the next section. Sticky trap counts or beat tray counts of adult pear psylla are positively correlated with percentage of growing terminals infested with nymphs or eggs (Adams and Los 1989). Adult counts were also correlated with percentage of fruit blemished by sooty mold (Adams and Los 1989). Savinelli and Tetrault (1984) showed that nymphal counts predicted honeydew quantities on foliage during some portions of the growing season.

Horton (1994; see also Horton and Lewis 1997) used linear regression to compare estimates of adult psylla densities obtained by sticky traps, beat trays, and whole branch samples (using large mesh bags; see above). Counts were linearly related among the three methods, but showed substantial unexplained variation around the regression line (Horton and Lewis 1997). Moreover, for any pair of methods, both regression slope and intercept varied with orchard, time-of-year, psylla density, sex, and psylla morphotype, suggesting that the linear relationship between methods was not constant. Other variables potentially affecting the regression lines may have included weather, pest management practices, phenological stage of the tree, or psylla behavior (discussed in detail below). The most consistent regression lines among orchards was the relationship between beat tray counts and sticky trap counts for the autumn winterform population (Horton and Lewis 1997). An absence of pest control in autumn may have contributed to this constancy.

Density vs activity: What is being monitored by sticky traps?

Horton (1994) showed that absolute density (numbers per leaf) of summerform adults was better predicted using beat trays than sticky traps, and concluded that sticky trap counts of pear psylla often included a large behavioral or flight activity component that obscured density effects. These two factors (density and activity) are often difficult to separate in the field using monitoring methods that trap flying insects (Williams 1940, 1951, Johnson 1969), thus caution is necessary when using sticky trap counts to make inferences about absolute densities of insects (Pienkowski and Medler 1966, Horton and Lewis 1997). In sum, unless they are quantified, factors that affect flight tendencies of pear psylla can obscure the true relationship between absolute density and trap count, potentially leading to erroneous estimates of damage potential.

The importance of flight activity by pear psylla in affecting counts on yellow or clear sticky traps has been explored in some detail (Horton 1994, Horton and Lewis 1997). By simultaneously monitoring pear psylla with sticky traps and beat trays, these authors were able to relate sticky trap catch to beat tray counts using linear regression. The authors then assumed that tray counts provide a fairly good quantitative index of absolute densities (Horton 1994) – at least within a single generation – and then further assumed that variation not explained by the regression was due to flight activity (plus experimental error). An examination of residuals from the regression line then provides some clues about patterns in activity under field conditions.

Factors affecting flight activity in psylla included physiological status of the insect, sex of the insect, and environmental conditions (Table 4). Summerforms showed two fairly distinct periods of high activity separated by a period of relatively low activity (Horton 1994). Dissections of female psylla during these different intervals indicated that periods of low activity occurred when a large proportion of the females in the population had immature ovaries (i.e., had only recently eclosed), suggesting that young psylla tend not to fly as much as older adults; Horton (1994) hypothesized that the younger adults also had somewhat immature flight muscles. Thus, sticky trap counts would underestimate absolute densities of psylla during intervals in which new adults were appearing compared to estimates made later in the generation when adults were older. Or, to state this differently, for a given absolute density of adults, sticky trap counts would be higher during a sampling interval in which a large proportion of adults were mature than during an interval in which most adults had recently eclosed. Other non-density factors potentially affecting counts of summerforms on sticky traps include sex of the insect, time of day, and location of the trap in the canopy (Table 4).

Seasonal changes in behavior also complicate the interpretation of sticky trap counts. For example, male winterforms have been shown to be more active than females during the reproductive period (spring) but not during the autumn diapause period (Horton and Lewis 1997). The change in behavior apparently is due to search for mates by reproductive males in the spring (pear psylla that are in diapause mate only very infrequently; Krysan and Higbee 1990). This shift in activity obviously obscures the relationship between absolute density of winterform psylla and density as estimated by sticky traps. That is, for a given beat tray count,

Table 4. Changes in flight activity shown or inferred to be important in affecting magnitude of counts on sticky traps

Response	Reference
Summerforms	
Lower activity levels at the beginning of a generation (young adults) than at mid-generation (mature adults)	Horton (1994), Horton and Lewis (1997)
Higher activity levels by both sexes in upper canopy than in lower canopy	Horton (1994)
Higher activity levels for males than females	Horton(1994)
Higher activity levels in midmorning than at other times	Horton (1993)
Spring (post-diapause) winterforms	
Increased activity in males between March and May	Horton and Lewis (1997)
Increased attraction to yellow by both sexes between March and May	Krysan and Horton (1991), Horton and Lewis (1997)
Higher activity levels for males than females	Horton (1993), Horton and Lewis (1997)
Higher activity levels at midday than at other times	Horton (1993)
Fall (diapause) winterforms	
Highest levels of activity during period of leaf fall in pear	Horton et al. (1994b), Horton and Lewis (1997)
Higher levels of activity during warm, sunny conditions than during cool, cloudy conditions	Horton et al. (1994b)

sticky traps potentially would overestimate male densities in spring (relative to estimates made in fall), and would also overestimate male densities relative to female densities in spring. Furthermore, activity by males in spring was not constant over the duration of the sampling period. Male flight activity increased between March and May (Table 4), apparently because densities of females declined in absolute terms over this same period. Low densities of females prompted dispersal by reproductive winterform males in small cage studies (Horton and Lewis 1995), and a similar response by males in the field would help explain patterns noted during the spring in the study by Horton and Lewis (1997). Finally, winterform psylla exhibit a seasonal change in response to yellow (Table 4). Winterforms of both sexes are actually somewhat repelled by yellow in February and March, and only become highly attracted by yellow once foliage begins to show in the pear tree (Krysan and Horton 1991). Thus, yellow sticky trap counts from a February sample would underestimate absolute densities relative to counts made in April if one were to assume a constant relationship between trap catch and tray counts.

Environmental conditions such as temperature almost certainly affect sticky trap counts of insects by influencing flight tendencies (Williams 1940, Johnson 1969). There is some indication that warm temperatures prompt flight in autumn populations of winterform pear psylla (Table 4). In a long-term sampling study, Horton et al. (1992) suggested that dispersal rates (numbers leaving the orchard) were lower during autumns that were wet and cool than in years that were warm and sunny. These authors also suggested that densities the following spring were higher following an autumn of reduced dispersal rates. A second environmental factor affecting pear psylla flight is leaf fall in pear (Table 4). Sticky trap counts increase substantially during leaf fall in pear, despite constant or decreasing (due to emigration) winterform densities during this interval (Horton et al. 1993, Horton et al. 1994b). Horton et al. (1994b) showed that leaf fall in pear dislodged pear psylla from the tree, and suggested that a large portion of the yellow trap catch at that time of year was due to dislodged psylla attempting to recolonize the pear tree.

In sum, research described in this section suggests that sticky trap counts of pear psylla are a function of (at a minimum) density, sex, morphotype, adult maturity, time of year, time of day, diapause status, sex ratio, leaf fall, and weather (Table 4). Decisions as to what monitoring tool should be used in a pest management program for pear psylla must consider that sticky trap counts will include both density and behavioral components. Because of this, beat trays and spur/leaf samples may be more useful than sticky traps for use in pest management programs, although the latter tool should not be immediately dismissed and can in fact provide useful information in control programs for pear psylla (Adams and Los 1989, Horton and Lewis 1997).

Conclusions

Difficulties in controlling pear psylla make it essential that an efficient monitoring program is a part of any pest management program for this species. A variety of methods have been developed to meet this need. The tools most commonly used for making pest management decisions are beat trays, used to monitor adults, and visual inspection of spurs and foliage to monitor eggs and nymphs. Some quantitative information is available describing the relationship between densities of pear psylla and damage to pear fruit caused by honeydew (i.e., russet), allowing rough approximations of action thresholds to be made for tray and foliage counts. Uncertainties remain, however, as to the extent that thresholds depend upon time of year, type of injury to be prevented, and pear cultivar, and these uncertainties have somewhat hampered development of a general sampling protocol for pest management purposes. For example, a sequential sampling plan has yet to be developed for spur and foliage samples, due to uncertainties about action thresholds coupled with a lack of information about spatial distribution of eggs and nymphs.

A third type of tool commonly used in monitoring pear psylla is the sticky trap, and a number of studies have made use of this monitoring tool to answer some specific questions in research. One important conclusion from this research is that sticky traps may be less useful than trays or foliage samples for use in pest management programs due to difficulties in interpreting trap counts. That is, counts of pear psylla on traps are a function of both density and flight activity. The latter depends upon a number of factors that are quite independent of density, thus counts of pear psylla on sticky traps potentially could lead to erroneous conclusions about densities and potential for impact.

I conclude this review with some brief suggestions about research that would improve our abilities to monitor (hence, manage) pear psylla:

1. What is the spatial distribution of adults, nymphs, and eggs as estimated using beat trays or

foliage samples? Does spatial distribution change seasonally?

2. How does control program affect the action threshold? More specifically, how does the change from a prebloom program that emphasizes use of adulticides to one that relies upon ovicides alter the existing thresholds?
3. Can we develop treatment thresholds based upon the combined densities of different life stages? If both adults and nymphs are just below their respective thresholds, is the combined density nevertheless high enough to cause economic damage?
4. What are the quantitative relationships between samples obtained at different times of the year? For example, a tray count of (say) 20 in February, when trees have no foliage, almost certainly indicates a different absolute density (number per tree) than a count of 20 obtained in mid-summer. There has been almost no research conducted that addresses this question.
5. Finally, can we develop a sequential sampling plan based upon presence/absence of eggs and nymphs, rather than having to estimate actual numbers per sampling unit? The availability of this type of plan would substantially reduce the time and labor necessary to process foliage samples.

Acknowledgements

I thank Carrol Calkins, Brad Higbee, Tom Unruh, and (especially) Rick Hilton for comments on an earlier draft of this paper. I also thank Tamera Lewis for her expert assistance in the field. My research on pear psylla has been generously supported by the Washington State Tree Fruit Research Commission and The Winter Pear Control Committee.

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