SAN JOSE SCALE AND ITS NATURAL ENEMIES: 
INVESTIGATING NATURAL OR AUGMENTED 
CONTROLS

PROJECT LEADER: Dr. Kent M. Daane
COOPERATORS: Glenn Y. Yokota, Walter J. Bentley, Karen Sime, and Kevin Fingerman

ABSTRACT

The biological control laboratory has studied San Jose scale (SJS) and its natural enemies from 1999 through 2003. This past year, our research focused on (1) completion of *Aphytis vandenboschi* laboratory studies, (2) initiation of *Encarsia perniciosi* laboratory studies, (3) determining seasonal SJS and natural enemy phenology, and (4) investigating a fall-season mass release of *Aphytis vandenboschi*.

The most important SJS parasitoids are the small wasps *Encarsia perniciosi*, *Aphytis aonidiae*, and *Aphytis vandenboschi*. Previous work showed *Encarsia* was the dominant parasitoid, although the *Aphytis* species are a critical component of biological control, especially when SJS populations reach higher densities. While these parasitoid species are found in most orchards, their effectiveness is variable and difficult to predict. Therefore, we conducted laboratory studies of parasitoid biology to better understand their strengths and limitations as SJS biological control agents. Studies with *A. vandenboschi* showed it uses second and third instar SJS for oviposition (depositing an egg), while first instars (crawlers) and male SJS are more typically used for “host feeding,” which often kills more SJS than through parasitism. *Aphytis vandenboschi* temperature development was studied at constant temperatures between 12 and 36°C (54 to 97°F). Parasitoid development was a significant linear function of temperature between 18 to 32°C (the higher the temperature the faster the parasitoid developed). However, below or above this range, development slowed and, at the extreme temperatures tested (12 and 36°C), there was significant parasitoid mortality. The results suggest that *Aphytis vandenboschi* is an excellent SJS natural enemy when it has suitable SJS stages (second and third instars) available and air temperatures range from 55 to 90°F. We suspect its limitations result primarily from summer periods of asynchrony with SJS host stage availability (when only very small SJS are found) and higher summer temperature intolerances.

We have successfully established a colony of *Encarsia perniciosi* and we are currently conducting host stage preference and temperature studies, similar to those conducted with *A. vandenboschi*. These trials are not yet completed.

Our field research component has been reduced and refocused. In previous studies we sampled many different orchards to provide an overview of SJS and parasitoid densities. This past year
we focused on three orchards, with SJS collected monthly and dissected to determine SJS stage and condition and parasitoid stage and condition. This year-long project is in its seventh month and collected data have not yet been analyzed.

Over the past three years we have investigated the potential of mass releases of *Aphytis vandenboschi*. These studies were conducted in small cages because we could not produce the needed number of parasitoids for larger-scale trials. This past year, our efforts have been joined by the Foothill Agricultural Research insectary. They have produced 1000s of *A. vandenboschi*, which have been field-released at three different sites (Kearney Agricultural Center and two commercial orchards). Because laboratory trials indicate that mid-summer air temperatures may reduce release effectiveness, we concentrated our release trials in the late summer and fall (20 August 2003 to present). Pre-release samples were collected during the spring and summer months. Post release samples will be collected beginning in December and continuing through harvest 2004. These trials are not yet completed.

INTRODUCTION

San Jose scale, *Diaspidiotus* (formerly *Quadraspidiotus*) *perniciosus* (Comstock), is a “hard” or “diaspid” scale. It is so small that its first development stage is often hard to see on fruit or branches. It has a large range of susceptible host plants that include stone fruit, pears, apples and many nut crops (Gentile and Summers 1958, see UC IPM website). It is most likely of Asian origin, brought into California in the 1870s on peach trees shipped from China (Gentile and Summers 1958). Because it has wide geographic and host ranges, it quickly became a key pest of most deciduous fruit orchards in North America and remained so until an Integrated Pest Management (IPM) program was developed, based largely on insecticide controls. Much of the development work towards a SJS program was completed in California.

From the 1950s-90s, when SJS populations flared up, control was often easily achieved through a well-timed insecticide application of a dormant oil, which was typically combined with an organophosphate, or spring and summer applications of organophosphates (Dowing and Logan 1977, Westigard 1977, 1979, Rice et al. 1979). The dormant season oil and organophosphate application targeting peach twig borer, *Anarsia lineatella*, also provided SJS control. More recently, the use of higher grade oils or bacterial-by-products (Success®) offered promise for spring and summer suppression of SJS. Walt Bentley and Rich Coviello are working on improved controls with oils and other products (W. J. Bentley, pers. comm.).

Because insecticides work best against the smaller scale, insecticide applications are often timed to periods of peak crawler emergence. Phenology models of SJS development have been used to improve application timing. Because the crawlers are hard to monitor and count, their emergence patterns are best determined based on “phenology” or development models that use the adult male flights to fix important periods in the SJS development patterns (Jorgenson et al. 1981). Sampling for SJS utilizes pheromone traps that attract adult male SJS – the only development stage and sex that flies. SJS pheromone traps can also attract one of the primary SJS parasitoids – a small “aphelinid” wasp named *Encarsia perniciosi* (formerly called *Prospaltella perniciosi* Tower) (McClain et al. 1990, Rice and Jones 1982). Two other SJS
parasitoids, *Aphytis aonidiae* (Mercet) and *Aphytis vandenboschi* Rosen can also be found on the SJS pheromone traps.

The combination of good sampling methods, a phenology model to time insecticide applications, and reliable insecticide products has been the standard for most IPM programs. For the above reasons, it was unusual when high densities of SJS were reported in the 1990s on stone fruit and almonds throughout the Central Valley and in particular in Fresno, Tulare and Kern counties. This past year, SJS infestations have been reported in more northern counties and in almonds and walnuts (Nick Mills and Roger Duncan, pers. comm.). This is particularly unusual because most almond and stone fruit growers with farms in Merced, Modesto and Stanislaus counties have rarely had to apply treatments for SJS (except on nectarine cultivars on which SJS readily settle on the fruit and even small populations can result in noticeable cosmetic damage). The exact causes of these outbreaks are not known but insecticide resistance, insecticide disruption of SJS natural enemies, poor insecticide application methods (e.g., poor coverage), and natural between-season fluctuations have been questioned.

Here, we summarize 1999-2003 studies of (1) the population dynamics (or the change in density over time) of SJS and its natural enemies, (2) seasonal changes in SJS and parasitoid densities between orchards with different management strategies, (3) within tree distribution of SJS and its parasitoids, (4) parasitoid species density and effectiveness, (5) sampling efficiency, and (6) augmentation of parasitoid species. We will present key data sets and graphs, although data from all individual orchards is available. This work was supported by joint funding from California Tree Fruit Agreement, California Cling Peach Growers Advisory Board and the Almond Board of California.

**OBJECTIVES (1999-2003):**

1. To compare potential field and laboratory effectiveness of *Encarsia perniciosi*, *Aphytis aonidiae*, and *Aphytis vandenboschi* – three parasitoids of the San Jose scale.
2. To investigate *Aphytis vandenboschi* biology and determine San Jose scale host stage preference, levels of host feeding, temperature tolerances and potential for insectary rearing.
3. To investigate interactions between *Encarsia perniciosi*, *Aphytis aonidiae*, and *Aphytis vandenboschi*.
4. To survey parasitoid populations and their relative abundance in northern, central and southern San Joaquin Valley locations.

**PLANS AND PROCEDURES**

*Field Populations.* The densities of SJS and resident natural enemies were followed in stone fruit and almond blocks in Fresno, Tulare and Kern counties from 1999 until 2001. Sampled orchard blocks represented different types of management systems for insect pests, which we have loosely categorized as conventional or sustainable based on the presence or absence of a dormant oil and organophosphate treatment for SJS or other insect pests (Table 1). The goal was to follow SJS and natural enemy densities over more than one season in many different orchards systems to determine if “patterns” in SJS and natural enemy densities held true. For example, if natural enemy densities respond to SJS in a “density-dependent” manner, then high SJS densities
and damage in 1999 would be followed by increased natural enemy activity and lower scale density in the next generation or season. Furthermore, if natural enemies can predictably control SJS, than scale densities should not remain consistently high in orchards without insecticide treatments for SJS. Because these events may not occur in a single season, multiple study years in the same fields were employed. Similar data were also collected in almond orchards.

Table 1. Sampled fields, categorized as “sustainable” (S) or “conventional” (C) management practices based on the dormant and in-season treatments applied and year (1999-2001) during which sampling occurred for stone fruit orchards (almond orchards not shown)

<table>
<thead>
<tr>
<th>Management</th>
<th>Block &amp; Cultivar</th>
<th>Year</th>
<th>Dormant</th>
<th>In-Season</th>
</tr>
</thead>
<tbody>
<tr>
<td>C</td>
<td>Royal Glo Nectarine</td>
<td>1999-2000</td>
<td>Supracide/Oil</td>
<td>Lannate</td>
</tr>
<tr>
<td>C</td>
<td>Rose Diamond Nectarine</td>
<td>1999</td>
<td>Lorsban/Oil</td>
<td>Lannate</td>
</tr>
<tr>
<td></td>
<td></td>
<td>2000-2001</td>
<td>Lorsban/Oil</td>
<td>none</td>
</tr>
<tr>
<td>C</td>
<td>Spring Bright Nectarine</td>
<td>1999-2001</td>
<td>Lorsban/Oil</td>
<td>Carzol</td>
</tr>
<tr>
<td>C</td>
<td>Arctic Snow Nectarine</td>
<td>1999</td>
<td>Sevin/Oil</td>
<td>Pencap (2X)</td>
</tr>
<tr>
<td></td>
<td></td>
<td>2000</td>
<td>Asana/Oil</td>
<td>Success</td>
</tr>
<tr>
<td>C</td>
<td>Honey Kist Nectarine</td>
<td>1999</td>
<td>Oil</td>
<td>Bt, Lannate</td>
</tr>
<tr>
<td></td>
<td></td>
<td>2000</td>
<td>Oil</td>
<td>Bt Carzol</td>
</tr>
<tr>
<td>S</td>
<td>Favorite Sun Nectarine</td>
<td>1999-2000</td>
<td>Oil</td>
<td>Bt</td>
</tr>
<tr>
<td></td>
<td></td>
<td>2000</td>
<td>Oil</td>
<td>Bt, Carzol</td>
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<tr>
<td></td>
<td></td>
<td>2001</td>
<td>Oil</td>
<td>Bt</td>
</tr>
<tr>
<td>S</td>
<td>Elegant Lady Peach</td>
<td>1999-2000</td>
<td>Oil</td>
<td>Bt</td>
</tr>
<tr>
<td>S</td>
<td>Friar Plums</td>
<td>1999-2000</td>
<td>Oil</td>
<td>none</td>
</tr>
<tr>
<td>S</td>
<td>Elephant Heart Plums</td>
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<td>Oil</td>
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</tr>
<tr>
<td></td>
<td></td>
<td>2001</td>
<td>none</td>
<td>none</td>
</tr>
<tr>
<td>S</td>
<td>Laroda Plums</td>
<td>1999-2000</td>
<td>Oil</td>
<td>Bt</td>
</tr>
<tr>
<td>S</td>
<td>May Grand Nectarine</td>
<td>1999-2001</td>
<td>Oil</td>
<td>Bt</td>
</tr>
</tbody>
</table>

In each orchard, 3-5 pheromone traps were used to sample for male SJS. In 1999, sampling began in April; in 2000 and 2001, we initiated sampling in March to catch the beginning of both the SJS and parasitoid flight activity. Cards were collected and lures replaced every 1-3 weeks, depending on seasonal SJS activity. In the beginning of the 2001 season, we increased sampling periodicity to 1-3 days to determine the relationship of male SJS and Encarsia perniciosi adult flights and density patterns.

Pheromone trap counts of SJS males also represent the SJS population development because the male flight corresponds with periods of maturing female SJS, such that adult male SJS flights are synchronized with female SJS in development stages receptive for copulation and the egg maturation process. To determine how long after the SJS adult male flight that SJS crawlers are found and whether or not pheromone trap counts could be correlated with the density of crawlers emerging from adult females, we placed double-sided sticky-tape around two limbs of each tree with a pheromone trap. Traps and tape were changed weekly from April (1999) or March (2000) until December.
At harvest we collected and examined 100 fruit in each of ten trees in each orchard. The number of infested fruit with either SJS, worms, katydid, and thrips damage; and the number of SJS per fruit were recorded.

Parasitoids can be reared from SJS on limbs, branches or fruit, and we periodically took infested wood and fruit to the laboratory specifically to rear out parasitoids. However, these samples may not represent natural enemy density relative to SJS in all orchards because it is quite labor intensive to find enough infested wood when SJS densities are low (e.g., in those orchards where natural enemies may be most efficient). For this reason, we also placed squash infested with SJS (from the insectary) in selected orchards to create a “controlled SJS population” at different times of the year. To accomplish this, squash were infested with ~1000 SJS, after the scale settled (once settled the scale will not move) and developed to host stages acceptable to the parasitoids (2nd and 3rd instars) the squash were placed in orchards. We used an open-topped plastic container, supported from a limb by metal hangers, to hang the squash in the tree canopy so that the squash did not contact tree limbs and parasitoids had to fly onto the squash to attack the scale. The squash were left in the orchard for 2-3 weeks, returned to the laboratory, and held in parasitoid emergence containers for up to 1 month. The emergence containers were checked weekly for adult parasitoids drawn into Dixie cups through a light trap, all collected parasitoids were identified to species. We used the “SJS-infested squash” to sample for parasitoids during summer and fall, 1999-2001. Additionally, in 2001 we used SJS-infested stone fruit seedlings to compare the number of emerged parasitoids with that emerging from SJS-infested squash.

**SJS and Parasitoid Distribution.** Where in the canopy SJS and its parasitoids are located can have important implications for trap placement, insecticide control and canopy management. In 2000 and 2001, we compared parasitoid and SJS distribution in the canopy by placing 3 × 5 inch sticky cards in different locations. Three treatment comparisons were tested: (1) high or low, (2) inner or outer, and (3) north, east, south or west directions. There were five trees (replicates) used for each treatment comparison, with cards left in the trees for 4 weeks, after which, all SJS and parasitoids were recorded. Each replicated trial was conducted in three stone fruit blocks (Laroda plums, May Grand nectarines and Sweet Home nectarines) and at three different periods of the season (March, July and September) and was repeated in March, July and September, placing cards in the orchards during periods of peak parasitoid emergence during the 1st, 2nd and 3rd – 4th SJS flight periods.

Using the same format, we also tested SJS and parasitoid species attraction to (1) yellow or white sticky cards and (2) white cards with or without a SJS pheromone lure.

In 2000 and 2001, we continued to study SJS and parasitoid distribution by destructively sampling entire scaffolding branches (2000-01) or spurs (2001). For scaffolding branches, a 15-year old May Grand nectarine block with high SJS densities was used. On each of three trees, one scaffolding branch was cut at the base and the entire scaffolding and its off-shoots were divided into upper or lower and inner or outer sections of scaffolding, fruiting wood (or “hangers”), new growth, sucker wood and leaves. Spur samples were collected in seven stone fruit orchards and three almond orchards in spring 2002. We collected 10-20 spurs on each tree, at the beginning of the season, and in some orchards we recorded spur location as we had done
with sticky cards. The number of spurs collected in each orchard varied with SJS density, in total, nearly 3000 spurs collected and dissected. For both scaffolding and spur samples, the collected material was bagged and stored at 34°F until dissected, recording SJS by stage and condition (live, dead, parasitized). From the presence or absence of a parasite exit hole, underneath the hard scale covering, the parasitoid species could be identified as either Encarsia, which has an emergence hole on only the outer carapace, or Aphytis, which has an emergence hole on the outer carapace and the SJS body.

To determine the kinds and numbers of SJS predators, beating-tray samples were completed on 20 trees per orchard per month during the growing season.

**Augmentation of Parasitoids.** In 1999 we tested the potential of a commercially available parasitoid (Aphytis melinus) to attack SJS in the field. Initial laboratory studies show this parasitoid species would host feed and parasitize SJS. However, tests in the open-field found this species did not affect SJS density or parasitism levels. For this reason, we established insectary colonies of SJS parasitoid: Aphytis vandenboschi (1999-present), Encarsia perniciosi (2000 and 2002) and Aphytis aonidiae (2002). Material from these colonies was used for (1) field augmentation trials, (2) bioassays of commonly used insecticides, and (3) laboratory studies of parasitoid biology.

In 2000 and 2002, we tested the effectiveness of mass-releases of Encarsia perniciosi and Aphytis vandenboschi. The experimental block was located at KAC and had not received insecticides for the previous 10 years. Earlier samples indicated that SJS, Encarsia perniciosi, Aphytis vandenboschi and Aphytis aonidiae were present in the orchard. To begin, selected branches were checked for SJS that, if found, were removed. After which, selected branches were inoculated with ~500 SJS crawlers (from the insectary) in late spring or summer to match the natural SJS population. The inoculated branches were enclosed in large, self-supporting organdy cages (~1 m long × 0.4 m diameter). After the SJS reached an appropriate size, a pre-release count of the number of settled SJS was determined for each branch. Encarsia perniciosi or Aphytis vandenboschi were then released into the cages at either 1:5 or 1:10 ratios of parasitoid : SJS (2000) or a 1 : 20 ratio (2002), based on pre-release counts. After 4-6 weeks, the cages were removed, the branches were cut from the trees and taken to the laboratory, and SJS found were counted and dissected to determine their development stage and condition (live, dead or parasitized).

In a second augmentation trial, the same orchard block at KAC was used for an “open-field” release. From 31 July to 3 August 2001, four branches on each of six trees were inoculated with ~200 SJS crawlers. On 25 August a pre-release count was made and on 1 and 6 September, 50 Encarsia perniciosi and Aphytis vandenboschi were released in three randomly selected trees. On 21 September the branches were removed and the SJS condition on each branch was recorded as before.

Results from these earlier trials showed the parasitoids had some promise, but could not realistically control a “SJS outbreak. For this reason, 2003 research focused on late summer and fall releases of A. vandenboschi, for use as an inoculative control of SJS the following season. Release rates and methods are as described above for the open field release, but the releases
began in August 2003 and the SJS density and parasitism levels will not be measured until spring 2004.

**Insecticide BioAssays.** In 2000, the effect of insecticide applications that target insect pests other than SJS, such as peach twig borer and thrips, on selected natural enemies common in almond and stone fruit orchard was evaluated in an almond block at KAC. Insecticide residue tests were conducted on both targeted pests (peach twig borer, oriental fruit moth and the navel orangeworm) and common beneficial insects (*Aphytis vandenboschi*, *Encarsia perniciosi*, *Stethorus* sp., golden eyed green lacewing, and *Goniozus legneri*). Results showed all material tested had a significant impact on the moth pest species. Our primary goal was to ascertain any differential impact on selected parasitoid, and results in 2000 show that some of these soft materials can have a negative impact on beneficial insects. These results were called into question and, for that reason, we repeated trials in 2002 with selected natural enemy species.

To begin, individual almond trees were hand-sprayed with common “soft” insecticides (Dimilin, Success, or Confirm) at label rates. Trees were set in a randomized complete block design (3 blocks), with 5 trees separating insecticide treatments within each row and 3 rows separating each block. Control trees received a water-spray. At 1, 7, and 14 days after insecticides were applied, nuts were collected from each tree and taken to the laboratory where they were examined to make sure that none of the tested nuts had insects or insect damage. Nuts were placed individually in small, ventilated containers, tested insects were added, and the containers were placed in a ventilated hood to reduce any fuming action of the insecticides in the containers. Insect condition (live or dead) was checked 2-3 days thereafter. In 2001, we tested *Aphytis vandenboschi*, golden eyed green lacewing, and *Goniozus legneri*. There were 20 replicates for each tested insect species and post-application period (1, 7, and 14 days).

**Parasitoid Biology.** In 2001 and 2002, laboratory work focused on the biology of *Aphytis vandenboschi*. In 2003, we completed research on *A. vandenboshi*, which proved proved the easiest to rear and was also a likely candidate for augmentative release. In fact either *Aphytis* species represents a suitable choice for an early-season release; resident *Encarsia* numbers are typically very high in the field at that time and do not warrant augmentation.

We wished to study the biology of *E. perniciosi* nonetheless in order to determine 1) why it falters later in the season, and whether augmentation would be of use then; and 2) whether it competes with the *Aphytis* species and thus might lower their effectiveness if combined in a release program. The major obstacle to the completion of this work has been the difficulty of establishing an *Encarsia* colony in the Berkeley insectary. In July 2003, however, we managed to rear the offspring of about 50 *E. perniciosi* originally reared from scales collected from almond, peach, and nectarine trees in Fresno and Kern counties. A portion of these offspring went into the competition experiment (below) and the rest into colony maintenance. By January 2004, we were consistently rearing several hundred *E. perniciosi* each week, enough to begin a series of biological studies.

Both *Aphytis* and *Encarsia* were reared on San Jose scales feeding on butternut squash. Squash were cleaned with 10% bleach, tripled rinsed, and then allowed to dry before SJS inoculation. SJS crawlers were collected from squash infested with gravid SJS by brushing the surface with a
fine-haired paint-blending brush, which was then tapped over the new squash to distribute the SJS crawlers. SJS and parasitoid “colony” squash are inoculated with 1000s of SJS crawlers each, while SJS and parasitoid “experimental” squash are inoculated with 100-300 SJS crawlers each. For *Aphytis vandenboschi* colonies, inoculated squash were held for 3-4 weeks (at 80°F) until most of the SJS reached the late 2nd to early 3rd instar. (Older scales were used because similar *Aphytis* species are reported to prefer this stage for oviposition (e.g., Luck and Podoler 1985).) At that time, the infested squash were placed in individual 1-gallon plastic containers and *A. vandenboschi* adults were added at a ratio of roughly 1:20 parasitoids:scales. After 20 days, *A. vandenboschi* adults began to emerge and were collected by attracting adults through a small funnel and into a glass vial by drawing them up toward the ceiling lights. The vials containing wasps could then be used in experiments or used to attack scales on another “colony” squash. Typically, two *A. vandenboschi* generations were produced from each squash (about 3 weeks apart). *Encarsia perniciosi* were reared in the same way except that they were given squash with younger scales, mostly 1st and 2nd instars (Yu et al. 1990). The cultures of the two wasp species were kept in separate rooms to prevent cross-contamination.

In 2003, we determined the host-stage preference for oviposition and host-feeding of *A. vandenboschi*, finished studying the effect of temperature on its development time, and began studies of its fecundity and of the effects of temperature on adult longevity. The effect of constant rearing temperatures was determined at 13, 15, 17, 20, 25, 30, 32, 33, and 34°C. For these experiments, squash bearing a mixture of scale life-stages (prepared as described above but with crawlers added over the course of 7-10 d) were exposed in 1-gallon plastic containers to approximately 50 *A. vandenboschi* adults. After 24 h, the wasps were removed. The infested squash were examined every 3 d for the higher temperature treatments (20°C and above) and every 5 d for the lower temperatures (13, 15, and 17°C). The scales were checked by flipping their covers with forceps beneath a dissecting microscope. Scale stage, condition, presence of host-feeding marks, as well as parasitoid stage when present, were recorded. Data from the first day of each trial were used to determine the host-stage preference for oviposition and host-feeding. Examinations continued until all of the *A. vandenboschi* had either died or emerged as adults. There were four replicates (each inoculated squash was a replicate) for each temperature treatment. Temperatures were maintained at T ± 1°C, with a 16:8 (L:D) photoperiod. Temperature cabinets were randomly assigned to each temperature treatment. Conditions inside the cabinets were monitored with Hobo™ data recorders (Onset Computer Corp., Massachusetts USA).

To determine the effects of temperature on the longevity of adult *A. vandenboschi*, wasps not more than 4 h old were collected from the colony and transferred to 40-mL glass vials provisioned with a streak of 50% honey-water. The honey-water was streaked on the top of the vial (when laid on its side) so that the wasps would be less likely to become stuck in it and die prematurely. These vials were then randomly assigned in groups of four replicates (each vial containing five wasps was a replicate) to five temperature treatments (15, 19, 23, 26, and 31°C) in cabinets prepared as described above. The vials were checked daily and the number of living and dead wasps recorded until all of the wasps in the treatment had died. The honey-water streaks were replenished when the vials were checked.
To determine lifetime fecundity, *A. vandenboschi* individuals were isolated beneath 35-mL plastic cups affixed with modeling clay to an area of squash surface infested with at least 30 scales. The wasps were provisioned with 50% honey water in streaks on the cup sides and thus had access both to a sugar source and to hosts. The cups containing the parasitoid individuals were moved to new locations on the squash surface, or to new infested squash if space was limiting, every 24 hours for the lifetime of the wasp. For the first treatment, the wasps were provided only 3rd–instar scales for the entire 24 hours of every treatment day. The second treatment allowed the parasitoid four hours of access to first instar scale by moving the cup with the wasp in it from the original squash to another squash infested with only first instars for the final four hours of each treatment day. For the third treatment, the wasps had access to third instar scale for only four hours, and spent the other 20 hours on a squash with first instar scale. The fourth and final treatment only allowed the parasitoid access to first instar scales for the full 24 hours. Upon the death of the parasitoid, the patches of scale that had been exposed to her were examined as described above for host feeding and oviposition. Each treatment was to be replicated with ten wasps.

A possible explanation for the different impacts of *A. vandenboschi* and *E. perniciosi* on field populations of SJS is that they compete with each other, as has been reported with *E. perniciosi* and other *Aphytis* species (Yu et al. 1990). Our first approach to determining whether and how competition might occur was to determine whether *A. vandenboschi* prefer or are repelled by SJS previously parasitized by *E. perniciosi*. We first exposed a squash bearing mainly 2nd instars (about 100 scales) to 20 *E. perniciosi* for 48 h. About half of the scale population on the squash was excluded from the wasps by a layer of plastic wrap. When most of the scales reached the 3rd instar (about 10 d later), they were exposed to *A. vandenboschi* for 48 h. The scales were then dissected, so that the numbers of *A. vandenboschi* eggs on scales exposed only to *A. vandenboschi* could be compared to the numbers on scales previously parasitized by *E. perniciosi*.

Our investigations of *E. perniciosi* biology are just now underway. The temperature-development studies have been conducted using the same methods described above for *A. vandenboschi* except we are only recording only times to pupation and adult emergence. Additional stage-specific data cannot be collected because *E. perniciosi* place their eggs inside the body of the scale and thus the eggs and larvae cannot readily be viewed. The adult-longevity studies have been conducted using procedures identical to those described above for *A. vandenboschi*.

**RESULTS**

**Field Populations.** Of the 11 stone fruit orchard blocks sampled there was a wide range of SJS density and damage. While we also recorded fruit damage from worms, katydid and birds – harvest samples indicate that damage from these pests was relatively low in most of the studied blocks during the sample periods (1999-2001). This is not surprising because we selected orchards based on their history of SJS density, not other pest species. What were the overall trends? Conventional orchards using a dormant treatment of oil and insecticide (Losban, Supracide, Asana or Sevin) or an in-season spray (Lannate or Pencap for moths, Carzol or Lannate for thrips) had low SJS infestations (<2%) in all 3 years studied (Fig. 1A). In those
blocks with only a dormant oil (without additional insecticides) and “soft” in-season treatments (Bt or pheromone confusion for moths, Success for thrips or katydids) there was a wide range of SJS fruit infestation (from 0-22%) during the three year period (Fig. 1B).

The general pattern would suggest that growers have very little control of their SJS pest densities if they choose to reduce or remove insecticide use. We believe this is not necessarily the case and will discuss general trends and individual orchard blocks to highlight SJS and natural enemy population density and damage. For example, there was a consistency in damage levels among years in the same orchard block studied. Simply stated, dirty fields stayed dirty and clean fields stayed clean (Fig. 1A). We had suspected that much of this difference might be explained by harvest date. In fact, there was some evidence that harvest date influences SJS fruit infestation levels in the conventionally managed fields (Fig. 1B; \( y = -4.6 + 0.03x, r^2 = 0.36, P = 0.02; 2 \) data points “□” were excluded from the regression analysis). However, we could not find a similar pattern of harvest date and damage in the sustainably managed fields \( (P = 0.78, 2 \) data points “■” were excluded from the regression analysis). Still, we would suggest that growers attempting to reduce insecticide use should first transition those blocks with the earliest harvest date to avoid most pest problems. The data presented on fruit infestation includes fruit with any amount of SJS – so a reported infestation rate of 1% may not have had any fruit culled (a few SJS, especially small SJS near the petiole, are not typically graded down).

We have searched for generalized trends to explain SJS and natural enemy abundance patterns and will present data from four years of field and laboratory studies. However, we should declare at the onset that for any generalized scheme developed, we are able to find exceptions – suggesting that each orchard and year can vary without a clear explanation.

A sticky card baited with a SJS pheromone lure is still one of the best methods to monitor the change in SJS density during the season. The cards provide information on SJS males (indicating peak flight periods and can be used to time insecticide sprays) and levels of *Encarsia perniciosi* – the most common SJS parasitoid. Figure 2 shows SJS pheromone catch data, from a conventional and sustainable field, and will be used to begin a discussion of the population
dynamics and biological control of SJS. In the conventional block, the grower used a dormant oil and an organophosphate for SJS and peach twig borer, and an in-season application of Lannate for thrips (which also provided some control of worms, SJS, and katydids). The sustainable grower used oil during the dormant season and controlled moth pests with either *Bacillus thuringiensis* (Bt), pheromones or, in some years, did not apply any summer treatments at all. These fields were selected because the collected data best fits our original hypotheses on SJS biological control (which has not held true across all blocks monitored).

In the conventional block, *Encarsia* adults are first collected in late March; soon after the *Encarsia* flight begins, the first SJS male flight occurs (from the overwintered population) but counts are very low as a result of the dormant treatment and not noticeable on the graph (Fig. 2). The second *Encarsia* flight peaks in May, before the second SJS flight, and is still quite high in comparison to any SJS numbers collected. The data suggest that *Encarsia* development is slightly faster than SJS, which will be discussed in greater detail. In late May or June, the second SJS flight occurs. This is an important flight because it provides the easiest count of the season’s SJS population. At the same time, counts of *Encarsia* have now decreased considerably, although there will be a peak in late June or July. In this block, SJS densities took a dramatic increase in the third SJS flight, in August, and again in the fourth SJS flight in mid to late September. During the third and most of the fourth SJS flight periods, the *Encarsia* density increases, but typically remains lower than the SJS counts. It is difficult to make out individual flights of *Encarsia*, indicating different generations, and by this time there is certainly an generations. In most stone fruit orchards, harvest has already occurred by these later flight periods, but these peaks can indicate next season’s SJS or *Encarsia* abundance.

In the sustainable block, they may outwardly appear to follow different seasonal patterns of SJS and *Encarsia*, as compared with those in the conventional block (Fig. 2), but, in fact, the seasonal pattern of both SJS and *Encarsia* flights are actually quite similar between the two fields. What is very different is the seasonal abundance of SJS and *Encarsia* in the two monitored sites and that difference given the impressions that there are different generation times. Most obvious is the high numbers of SJS and *Encarsia* in March. In most fields with high *Encarsia* densities there is a double peak, separated by about 3 weeks, seen in first (March/April) and second (April/May) *Encarsia* flights. It is unlikely, given March and April temperatures and *Encarsia*’s preference for 2nd instar SJS, that each peak represents a separate generation. More likely, the double peak represents the emergence of *Encarsia* that overwintered as a pupa and others that overwintered as larvae, leading to an off-set brood. Similar to the conventional block, the *Encarsia* numbers drop during the summer months and then increase in August and September. Unlike the conventional block, the fall flight of *Encarsia* had higher counts than SJS. The actual density of SJS in October was similar between the two blocks. The preferred overwintering stages of *Encarsia* (or *Aphytis* spp.) are not known and will be studied in winter 2002/03. Parasitoid numbers drop during the season and climb again, along with SJS numbers in the fall.

The generalized patterns for SJS and *Encarsia* presented in Figure 1 allow for a simple, but incomplete explanation. First, conventional wisdom suggests that insecticides applied in the dormant and early spring periods reduce SJS, which is correct, but also have a devastating impact on *Encarsia*, which is incorrect. Second, that sustainable fields should be managed to
support beneficial insects during the growing season to track SJS populations and result in consistently lower densities by the end of the season. This assumption may be incorrect or, at least, incomplete. Furthermore, it was incorrectly assumed that when parasitoids dominant the trap counts, as in the sustainable field, the SJS damage would be low.

We have selected six orchard blocks that represent both the consistent and, at times the chaotic, annual patterns of SJS and Encarsia abundance. From these examples, we will begin discussions of the potential strengths and weaknesses of SJS biological control.

SJS abundance varies among the conventional blocks (Fig. 3). In the “Rose Diamond” nectarine block, standard management practice included a dormant oil and Lorsban treatment, with only a single in-season treatment for thrips (Lannate – 1999). During each season, SJS abundance progressively increased; while there was a general decrease in overall abundance with each progressive year. Because SJS did not increase until late in the season, there was little fruit damage in this early harvested variety. Management practice in the “Spring Bright” nectarine block relied on a dormant oil and Lorsban treatment and an in-season Carzol treatment for thrips.
SJS density was consistently low. The third selected conventional block, the “Favorite Sun” nectarine, used only oil in the dormant application and an in-season application of Carzol for thrips when needed. This field had the greatest late season SJS increase of the conventional fields.

How important is *Encarsia* in the control of SJS in conventional blocks. Previously, we (and others) suggested that spring and summer *Encarsia* populations might be reduced by dormant or early-spring insecticide treatments of Lorsban, Lannate, or Carzol. The data suggests the *Encarsia* : SJS ratio is not consistently reduced by conventional management practices. In most fields there was an initially high *Encarsia* level compared with SJS. *Encarsia* is an internal SJS parasitoid that will cause the SJS body to “mummify” or harden underneath the SJS outer shell. This provides two layers of protection from the suffocating oil or the contact insecticide. In fact, the conventionally-managed “Spring Bight” nectarine block had one of the most consistently high, season-long presence of *Encarsia*.

SJS and *Encarsia* trap counts in sustainably-managed blocks followed a similar pattern to the conventional blocks – except the abundances of both pest and parasitoid were higher (Fig. 4). The initial ratio of *Encarsia* : SJS is higher in sustainably-managed blocks as compared with conventionally-managed blocks. However, viewing the pest and parasitoid abundance data over a number of years provides better insight into the seasonal abundance of *Encarsia* – which does not start in the spring but in the previous fall period. Large SJS populations at the end of the season result in an initially large *Encarsia* population the following year. The abundance of SJS males in the first flight (March/April) from sustainable blocks (in most years) and some conventional (“Favorite Sun”) blocks also show the importance and effectiveness of a well-timed dormant oil (a dormant oil was not used in the “Laroda” plum - 2001 and “Elephant Heart” nectarine – 2000) (Figs. 3 and 4). Still, blocks with a high first SJS flight had both high and low levels of fruit damage.
Fig. 3. SJS pheromone trap data for selected “conventional” blocks, 1999-2001.
Fig. 4. SJS pheromone trap data for selected “sustainable” blocks, 1999-2001.
Comparing *Encarsia* densities in conventional to sustainable blocks gives some indication of why *Encarsia* populations are not controlling SJS in all years and blocks. In almost every year and block, *Encarsia* numbers do not increase during the summer in response to the increase in SJS. We had previously believed that if SJS were present in the orchard, there would be a progressive increase in parasitoid densities, with *Encarsia* and *Aphytis* species having 2-3 generations for every SJS generation. This was based, in part on early season trap data that shows two *Encarsia* generations from March to June, during the time SJS has a single generation. We also had initial temperature development observation from the insectary colony that showed *Aphytis vandenboschi* developing from eggs to adults in about 2-3 weeks, while the SJS took about twice that time. At this 2:1 development rate of parasitoid : SJS, we hypothesized 8-10 generations of the parasitoids per year. However, observing the peaks and valleys of *Encarsia* during each season (Figs. 2, 3, 4) in order to estimate the number of *Encarsia* generations suggests only 5 *Encarsia* generations during each season – about the same number as SJS, which typically has 4-5 generations in the San Joaquin Valley.

Again looking at the seasonal density patterns of *Encarsia* (as measured by the sticky cards) suggests that there is a clear low during the summer months, with higher peaks in both the spring and fall periods. Yet to be decided is whether this pattern is a result of parasitoids responding to SJS densities or the parasitoids directly responding to summer temperatures.

The difference in SJS and *Encarsia* seasonal patterns can be clearly seen when the average SJS density – across all orchard blocks – is graphed for each year (Fig. 5A) and compared with the average *Encarsia* density – across all orchards blocks – also graphed for each year (Fig. 5B). SJS density begins low each season (Fig. 5A, there is a spike in the 1st flight in 2001 that was a result of two fields that did not have a dormant treatment). With each flight there was a general increase in SJS density, brought back down with the dormant treatment. In contrast, the *Encarsia* densities show a sharp decrease from the 1st flight in March/April through the 3rd flight in July/August (Fig. 5B). That the *Encarsia* population density is decreasing while the SJS density is increasing is not a good indication of the parasitoid’s potential to respond to SJS populations. The summer decrease in *Encarsia* is followed by a fall increase as the parasitoids
attack the overwintered SJS population. What causes the decrease? We believe there are two possibilities that should be further studied. First, that *Encarsia* (and perhaps *Aphytis*) have slower development and poor survival during the summer months because of high temperature intolerance. Second, that spring and summer SJS populations have periods where a majority of the SJS population is in development stages that are not preferred by *Encarsia* (or *Aphytis*) for oviposition.

**SJS and Parasitoid Distribution.** Pheromone-baited SJS sticky cards are still the easiest method to monitor SJS. Work in 1999 suggests that sampling methods for parasitoids need to be improved. Continuing this research in 2000-2001, we found that trap placement can affect recorded densities of SJS and we sought to determine the influence of trap placement on SJS : parasitoids ratios. Results from placement of sticky cards throughout the tree canopy (some with and others without pheromone lures) showed clear differences between tree canopy locations where adult male SJS and adult (female) *Encarsia* are more likely to be found. A short discussion and a few graphs of the collected data can summarize the influence of trap placement.

We will begin with a discussion of sticky cards *without* a SJS pheromone lure. Results show no difference in SJS or parasitoid (*Encarsia* or *Aphytis*) capture between cardinal directions (north, east, south or west) (Tukey’s HSD test, *P* < 0.05). Similarly, we found few differences in SJS or parasitoids collected on white or yellow sticky cards. Earlier work in North Carolina suggested that trap color (white, blue, black, yellow or red) influenced parasitoid catch (McClain et al. 1990). There were, however, important differences in sticky trap location with respect to the number of SJS or parasitoids collected. For example, results show that sticky cards placed in the inner canopy section generally caught significantly more parasitoids (8 of 9 trials) and SJS (7 of 9 trials) than sticky cards placed in the outer canopy sections. Results from the May Grand nectarine trial in March are presented (Fig. 6).

![Effect of Inner vs Outer Card Location Parasitoid and SJS May Grand Nectarines: March sample](image1)

![Effect of Inner vs Outer Position on the Distribution of Encarsia perniciosi to SJS](image2)

Fig. 6. The number of parasitoids and SJS caught on sticky cards (without a pheromone lure) placed in inner vs. outer canopy positions (data are shown for 1 of 9 trials).

Fig. 7. The ratio of parasitoids : SJS on sticky cards (without a pheromone lure) show significantly more parasitoids were caught in the inner vs. outer canopy positions during the March/April samples.
Remember that these trials were conducted using sticky cards WITHOUT a pheromone lure – which dramatically drops the number of *Encarsia perniciosi* and SJS that are caught (*E. perniciosi* is also attracted to the SJS pheromone [Rice and Jones 1982]). While both parasitoids and SJS were more frequently collected in the lower/inner portions of the canopy, the ratio of parasitoids to SJS becomes as important as the density. For example, will there be a predictable or successful level of biological if early season counts show a parasitoid to SJS ratio of 1000 to 1. This has important implications for sampling. Lower/inner card placement will describe a SJS and parasitoid complex with far more parasitoids than SJS. A higher/outer card placement will do just the opposite –leaving the PCA with the impression that there are far more SJS.

Results of parasitoid : SJS ratios in inner vs. outer canopy sections are presented separately for the three sampling periods (Fig. 7) and show that in March/April samples there were significantly higher parasitoid : SJS ratios. Note also that while *Encarsia* is more common than SJS in March samples, the ratio of *Encarsia* to SJS is 1 to 1 in July and September samples as the number of *Encarsia* is reduced during the summer months– as discussed previously.

![Graphs showing the ratio of parasitoids to SJS across different canopy sections for different collection periods.](image)

Fig. 8. Differences in the ratio of parasitoids : SJS on sticky cards placed in upper vs. lower canopy positions during three sampling periods (data from different orchards are combined in July/August and September/October sampling dates).
The same pattern shown with inner vs. outer sticky card placement was also found for upper vs. lower placement of sticky cards (Fig. 8.). Results are presented only for the ratio of parasitoids : SJS, and in these graphs we separate Encarsia from Aphytis because results show an interspecific difference. Altogether, results show that card placement can influence decisions made by PCAs and growers on the potential level of SJS biological control in the orchard. Furthermore, the data imply that more Encarsia are found in the lower/inner canopy sections.

Fig. 9. How does this information influence monitoring programs for SJS? The data presented have been from sticky cards without pheromone lures. This provides a better comparison of parasitoid and SJS in the canopy because the insects are not being “called in” by the SJS sex pheromone. The seasonal average shows that Encarsia, SJS, and Aphytis species are collected in relatively similar numbers. However, sampling methods would always make use of pheromone lures. The next series of graphs looks at SJS and parasitoid distribution with lures.

Fig. 10. The relationship between SJS and trap height held true when SJS pheromone lures were added. More SJS were collected in the upper canopy sections (Fig. 10A). In contrast, more Encarsia perniciosi (the only parasitoid species attracted to the SJS pheromone lure) were collected in the lower canopy sections (Fig. 10B). Combined, this relationship shows that trap placement in low canopy sections (which are easier to reach) will overestimate the Encarsia : SJS ratio and suggest more biological control agents are present than there actually are.
These results on differences in parasitoid and SJS distribution, as measured by sticky cards either with or without a sex pheromone lure, corroborate those 1999 studies that show differential distribution, with Encarsia perniciosi more common on the interior portions of the canopy and Aphytis spp. distributed throughout. Other researchers have suggested SJS is not evenly distributed – with SJS more common on the bark than on the leaves or fruit and on the smaller limbs on the tops of the tree than on the larger interior sections (Morgan and Angle 1969). However, this work was completed in British Columbia, Canada, where cultivars, seasonal temperatures, and natural enemies would be very different.

Can SJS and parasitoid distribution affect IPM programs? Yes! For example, insecticide application that provides better coverage to the outer portion of the tree – compared to the inner regions – might not provide the best SJS control and could negatively affect one parasitoid species (Aphytis) more than another (Encarsia). In some cases, poor insecticide coverage may be a function of the canopy shape and size. Older stone fruit orchards have thick, rough scaffolding branches where complete insecticide cover may be more difficult. Similarly, in older almond orchards the canopy can be quite tall and coverage in these upper regions is more difficult. Incomplete coverage in these situations would tend to favor Encarsia (which prefers the inner sections) more than Aphytis, which is distributed throughout the canopy.

From these results, we hypothesize that the more hidden location of Encarsia may provide some protection from insecticide applications as compared to the more exposed location of Aphytis. We also hypothesize that in “outbreak” seasons, both Encarsia and Aphytis species may be needed for natural control of SJS because different species may forage preferentially on different parts of the tree canopy. It has become apparent that the importance of Aphytis spp. has been sorely underestimated – probably due to the conspicuous presence of Encarsia on the pheromone traps. At the project’s conclusion, collected data will provide a better description of management practices that disrupt either SJS or its parasitoids and which may result in SJS outbreaks.

A complete description of SJS sampling should include a discussion of the use of double-sided sticky tape to monitor SJS crawlers. In Figure 11, we have taken the average counts from pheromone traps and sticky tape, taken in the same orchard, and placed both on the same graph. The relationship between pheromone and tape counts is quite clear. The season begins with a
small first flight of SJS adult males, captured by the pheromone traps, which is followed in about one month by an increase in the number of crawlers caught on the double sided sticky tape. The second SJS flight is similarly followed by an increase in the number of crawlers collected. On the graph (Fig. 11), the male flight and corresponding movement of crawlers from the next generation is indicated by “Comparison Set 1 and 2” for the first two SJS generations. Underneath the “X-axis” we have estimated the four SJS generations (not flights) apparent for the sampled orchards.

In summary, the data show a very good pattern of adult male catches in the pheromone traps followed by a corresponding increase in crawlers in the following generations. Can sticky tape be used to indicate potential economic injury levels? This idea was pioneered by W. J. Bentley (et al.). In their studies, they plotted sticky trap density against SJS damage at harvest and found a significant relationship, indicating the counts on double-sided sticky tape were related to damage. We also found this relationship to hold true (Fig. 12), when data were combined across all orchards and seasons.

![Comparison of pheromone traps to sticky tape catches](image)

Fig. 11. Seasonal averages across all fields for SJS males caught in SJS pheromone traps (●) and SJS crawlers caught on double-sided sticky tape (○) placed around branches on the same tree as the pheromone traps were hung (data are from 2000).
However, we believe that using sticky tape to predict damage is a poor replacement for pheromone traps for the following three reasons. First, the selection of which branches will be monitored can completely change the average counts because the tape will monitor crawler movement only on the monitored branch. Because the SJS is clumped, branches on the same tree can have sticky tape counts of 100s or only a few SJS crawlers. These results indicated that sample bias is a concern – do you put tape around limbs where you see SJS? It also suggests that to get an accurate count, 10s or 100s of limbs in each block must be sampled. In contrast, pheromone traps pull the SJS in from a distance and, therefore, only a few traps are needed for each block. Second, the crawler emergence follows SJS male flight activity. For this reason, the good crawler count in each season would be in late April to mid-May. Control decision may already have been made by that time and pheromone trap counts provide that information earlier in the season. Finally, a good correlation of tape counts to trap damage required a season-long average of sticky tape. As can be seen in Figure 11, the late season counts indicate a continual presence of SJS and are not as worthwhile to show peak activity periods or density.

We believe there is a very good purpose for the double-sided sticky tape – it should be used to determine egg hatch and crawler movement, not to indicate SJS density. If pheromone trap catches indicate a worrisome SJS population, then find branches infested with SJS and put sticky tape around those branches. Check the tape frequently to determine when crawler hatch begins and use this information to time the insecticide applications. This can be an important tool, especially for oil-only applications because the SJS crawler is the easiest of the SJS development stages to kill with oil applications.

**Augmentation of Parasitoids.** One of the more difficult projects has been the investigation of an augmentation program for SJS. The work described above - following SJS and parasitoid density in nine orchard blocks - brought one aspect of augmentation into better focus. It appears that mass producing and releasing *Encarsia perniciosi* early in the season would not be
beneficial. This parasitoid is in all orchards with SJS, regardless of insecticide use, and early season release would probably not be able to add significantly to parasitoid population.

One of the biggest hurdles for an augmentation program is the development of insectary procedures to mass-rear viable and effective natural enemies. As mentioned above, colonies of *A. vandenboschi* and *E. perniciosus* have been established. We are currently conducting laboratory studies on parasitoid preference for SJS stages – to better determine when to release the parasitoids – and parasitoid fecundity and host feeding limits – to better determine the release rates needed to suppress SJS.

However, our initial release studies were conducted with a limited number of available *Aphytis* and *Encarsia* from the colonies to release. In 2000, we compared *Encarsia* and *Aphytis vandenboschi*. Results show that *Aphytis*, while it may not parasitize as many SJS throughout the season, may kill considerably more due to the process of “host feeding,” which means the parasitoid sticks the SJS with its ovipositor to cause “bleeding.” The parasitoid feeds on the SJS juices to help develop eggs and the “poked” SJS eventually dies. Host feeding is far more common on small SJS, while larger SJS are used for egg deposition. Furthermore, initial collections of overwintered SJS indicate a greater presence of *Aphytis* than previously recorded.

This work was repeated in 2002, using a similar cage design but with a lower parasitoid : SJS release rate. In 2002, we released about 1 parasitoid (*Aphytis vandenboschi* or *Encarsia perniciosi*) for every 25 SJS. We also more closely separated out the release outcome in comparison to the development stage of SJS attacked. While the study was small, the data very closely follow out predictions, based on the previously discussed field studies. Data from both July and August trials are combined and we will discuss the two parasitoids species separately.

Results from *Encarsia* release show that 35.5% of 2nd instar SJS were parasitized, while only 20% of 3rd instar SJS were parasitized. There was very little host feeding, as indicated by the
number of dead SJS, although the smaller stage SJS had more dead scale (100% of 1st instars, 10% of 2nd instars and 6% of 3rd instars). We found no 1st instar female or male SJS were parasitized. We found only 10% of 2nd instar SJS were parasitized, while 42.5% of 3rd instar SJS were parasitized. There was more host feeding, with no live 1st instars found, 80% of the 2nd instars were dead, and 29% of 3rd instars were dead.

We believe these results are quite promising. A commercial insectary (Foothill Agricultural Research) is currently testing production methods for *Aphytis vandenboschi* and is supplying our laboratory with the needed material for our studies. Results from the 2003 fall releases will not be available until spring 2004.

**Insecticide BioAssays.** In 2000, we screened Dimilin®, Confirm®, and Success® for efficacy against peach twig borer (PTB). Dimilin and Confirm are insect growth regulators (IGRs), which are larvicides that interfere with the insects' chitin deposition (the outside shell) and this prevents the insect from molting. Success is a bacterial by-product and is currently registered for use in almonds. Our goal was to provide information that might better usher these “softer” products into widespread use. Because any insecticide application has the potential to affect other pest and beneficial insects in the orchard, we also screened the tested IGRs against some of the more common beneficial insects in almond orchards.

In 1998, and 1999, we used "diet-incorporated" bioassays to develop LD50s and showed that all three products were effective against PTB. In 1999, we began field tests of these products on PTB and some commonly found beneficial insects. In 2000, almond trees were treated, using commercial methodologies, with 2× the label rate for each product. Nuts from those treated trees and water treated control trees were collected at 1, 6, 12, and 22 days after spray application (as described for the 2002 study) and placed, individually, in plastic rearing cells. Our 2000 results suggested that green lacewings were not affected by any of the tested insecticides. We also found no discernable effect of the IGRs (Dimilin and Confirm) on *Goniozus legneri* or small SJS parasitoids (*Encarsia perniciosi* and *Aphytis vandenboschi*). However, there was increased mortality of the SJS parasitoids in the Success treatment. Because the mode of action of Success would suggest that this product should not kill adult parasitoids, and because there was considerable mortality in the control treatment for both *Encarsia perniciosi* and *Aphytis vandenboschi*, these results were held in question.

In 2002, we repeated the insecticide trials with green lacewings, *Goniozus legneri* and *Aphytis vandenboschi*. Nuts were collected at 1, 7 and 14 days post insecticide application (in 2002 insecticides were applied at label rates). Results from this past year’s trial are not ambiguous. Survivorship curves are presented for *Aphytis vandenboschi* (Fig. 15), *Goniozus legneri* (Fig. 16) and green lacewings (Fig. 17). Results show adult parasitoids in contact with nuts treated with Success had high mortality –100% of the 20 tested *Aphytis* were dead after one day, and this pattern held true in treatments 1, 7 and 14 days post application. There was also a significant, although not as dramatic, effect on the larger, more long lived *Goniozus*. In one trial (day 7) there was significant mortality of Confirm in the *Aphytis* treatment, although there were no effects of the IGRs on the larger *Goniozus*. 

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Other research, testing different beneficial insects, has shown that Success can increase mortality of parasitoids. We hypothesize that the residue on the almond nut is incorporated into the small moisture droplets that form on the nut – during morning dew, for example – and as the parasitoids feed on this moisture they are exposed to the insecticide.

In contrast, there was no effect of Success on green lacewings, but there was a significantly higher mortality of green lacewings in the Dimilin treatment. In these trials, the lacewing larvae were provided navel orangeworm larvae as a food source. We hypothesize that navel orangeworm, feeding on the almond nut, ingested some of the insecticide material, which was incorporated into the moth larva and then into the lacewing.

The research shows that these soft insecticides can still have some negative impact on the beneficial insect complex common in orchards. However, as in the 2000 report we emphasize that these products do an excellent job at controlling many insect pests; they have lower mammalian toxicity and lower broad spectrum effects against other animals (including insects) in the orchard ecosystem.
Fig. 15. The effect of insecticide residues from almond branches treated at 1, 7, and 14 days pre-exposure on *Aphytis vandenboschi* adults.
Fig. 16. The effect of insecticide residues from almond branches treated at 1, 7, and 14 days pre-exposure on *Goniozus legneri* adults.
Fig. 17. The effect of insecticide residues from almond branches treated at 1, 7, and 14 days pre-exposure on green lacewings larvae.
Parasitoid Biology. From 1999 – 2002, we used squash infested with SJS to compare parasitoid effectiveness and species composition at different times in the season. We reported previously that overall parasitism levels were often low. The ratio of parasitoids collected from these infested squash typically weighted towards *Encarsia perniciosi* (about 70% of the material collected) and *Aphytis aonidiae* and *A. vandenboschi* (about 30% of the material collected). We begin this report with a confession that our sampling methodology was flawed. We hung the infested squash in the trees, without any contact with the tree itself. In 2002, we also placed infested stone fruit seedlings in the canopy (courtesy of W. J. Bentley) with the plant material allowed to contact the tree itself. The parasitism rates from these infested seedlings was five times greater than the infested squash placed in the same tree for the same period of time. These results suggest that parasitoids use cues, such as the tree substrate on which the SJS are settled, to help find and determine whether or not to oviposit in the SJS. We bring this up only to point out how information on parasitoid biology is needed to better understand their effectiveness.

In 2002, results from laboratory trials began to provide valuable information on *Aphytis vandenboschi* biology. These studies are now for the most part complete. In 2003, we confirmed the upper and lower development thresholds for *A. vandenboschi* development. Graphed to a model fitting the data to a sigmoid curve with an upper temperature threshold (i.e., the slope of the curve drops as parasitoid development reaches its upper threshold) the results suggest that *A. vandenboschi* will not survive above 39ºC (Fig. 19). Temperature in the San Joaquin Valley often exceed this upper limit, however, and the data from the laboratory study are limited in that the study was conducted at constant temperatures. The results may have thus produced an artificially low theoretical limit, but they do strongly suggest that during the hot summer temperatures the *Aphytis* population will not develop well. We determined that optimal development occurs at about 28ºC.

The detailed results from the *A. vandenboschi* temperature-development study are presented in the bubble graphs below (Figures 19, 20). The X-axis is the sample date and the bubble over the X axis aligns on the Y axis with the parasitoid development stages found. The size of the bubble indicates the percentage of the parasitoid population found in each development stage. For example, at 15ºC all parasitoids found (in the dissected SJS) were in the egg stage until day 14, when we found 80% eggs and 20% 1st instar parasitoids. At 15ºC the *Aphytis* is developing, although at a very slow pace, and after 58 days most of the parasitoid population has reached the pupal stage. The population remained in this stage and these results suggest that, during the winter, the parasitoid may stay in the pupal stage until temperatures increase in spring. At 13ºC, development appeared to be disrupted. Very few adults emerged. Many wasps did not appear to pupate successfully, either remaining mature larvae or expelling meconial pellets without completing the pupal molt. At the other extreme, we identified a clear upper limit to development at 32ºC. At this temperature, most wasps developed normally, whereas at 33ºC, no wasps made it past the pupal stage, and at 34ºC, most wasps died as young larvae.

Comparable temperature-development studies for *E. perniciosi* are ongoing. Due to the (presumed) long development times at the lowest temperatures we do not expect to finish these studies until late April 2004.
The data have important implications for both natural and augmented biological control. For example, perhaps summer release of parasitoids would place insectary-reared material in the field at a time when environmental conditions are especially harsh for parasitoid survival. Prolonged periods of high summer temperatures would be expected to remove a larger percentage of the parasitoid population.

Fig. 18. *Aphytis vandenboschi* development at six constant temperatures – 15, 20, 25, 30, 32 and 33°C – show that the parasitoid has lower and upper temperature thresholds that would impair its development in some regions of the San Joaquin Valley.

Figure 20. Comparison of *A. vandenboschi* larval progress at the upper and lower thresholds of development.
Fig. 20. *Aphytis vandenboschi* development at five constant temperatures, with data presented in a “bubble” graph to depict parasitoid develop across time (explained in the discussion).
Our results for host-stage preference (Figure 21) confirm that *A. vandenboschi* prefers to lay eggs in 3rd-instar scales. Host-feeding, in contrast, concentrates on the younger stages. These results indicate that it is best to provide wasps with a mixture of scale stages to optimize rearing in the insectary, and that field releases should also be made to coincide with the availability of multiple stages. The relatively high incidence of host feeding—which almost always kills the host—indicate that the effect of this species on host populations is greater than parasitism rates would indicate alone, and thus further support its use in the field.

![Figure 21](image)

**Figure 21.** *A. vandenboschi* tend to lay eggs in 3rd-instar scales; 2nd instars are sometimes attacked but 1st instars almost never. They tend to host-feed on 1st and 2nd instar scales, rarely on older scales.

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**longevity overview**

![Figure 22](image)

**Figure 22.** Summary of results for adult longevity of *A. vandenboschi* with respect to temperature. At 31ºC, most wasps are dead after 3 d; at 15ºC, the wasps live for several weeks.
Our studies of *A. vandenboschi* adult longevity are now largely complete and reveal a likely upper limit to temperature tolerance at 31ºC (Figure 22). It will be of particular interest to compare these results to those for *E. perniciosi* (studies now in progress). Any differences between the two at higher temperatures would be extremely useful towards explaining their different field densities. We are now testing *A. vandenboschi* at 19ºC and will complete an additional trial at 34ºC to confirm its upper temperature limits. We began expect to finish the adult longevity experiments (for both *E. perniciosi* and *A. vandenboschi*) by early March 2004.

Our fecundity experiments are currently in progress.

The preliminary results of our competition study (only one squash-replicate has been completed to date) suggest that there is no behavioral discrimination by *A. vandenboschi* females among scales previously exposed to *E. perniciosi* females (Figure 23). If the completed experiment confirms this result, then we will conclude that *A. vandenboschi* do not actively interfere with *E. perniciosi* development, as would be suggested by the finding that they preferentially oviposit in scales previously parasitized by *E. perniciosi* (or vice versa, if *A. vandenboschi* were found to avoid scales parasitized by *E. perniciosi*). Instead, temperature or other physiological constraints would be more likely explanations for field performance. We do not yet have data on survivorship of either species in the two treatments. We expect to complete these experiments by June 2004.

![Figure 23. Percent parasitism by *A. vandenboschi* on areas of a squash not previously exposed to *E. perniciosi* (left) and heavily parasitized by *E. perniciosi* 10 d prior to exposure to *A. vandenboschi* (right). There is no difference in parasitism rates by *A. vandenboschi* between the two treatments, suggesting that *A. vandenboschi* do not discriminate between scales previously parasitized by *E. perniciosi*.](image-url)
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References

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