Biological Control **20,** 132–146 (2001)
doi:10.1006/bcon.2000.0886, available online at http://www.idealibrary.com on IDEAL®

Compatibility of Insect Growth Regulators with *Eretmocerus eremicus* (Hymenoptera: Aphelinidae) for Whitefly (Homoptera: Alyerodidae) Control on Poinsettias

II. Trials in Commercial Poinsettia Crops

R. G. Van Driesche,* M. S. Hoddle,† S. Lyon,* and J. P. Sanderson‡

**Department of Entomology, University of Massachusetts, Amherst, Massachusetts 01003; †Department of Entomology, University of California, Riverside, California 92521; and ‡Department of Entomology, Cornell University, Ithaca, New York 14853*

Received February 18, 2000; accepted September 9, 2000; published online December 12, 2000

The efficacy and cost of reduced release rates of the parasitoid *Eretmocerus eremicus* Rose and Zolnerowich (Hymenoptera: Aphelinidae) when combined with application of the insect growth regulator buprofezin were compared to those of a higher parasitoid release rate used alone for whitefly control (Homoptera: Aleyrodidae) on poinsettia (*Euphorbia pulcherrima* Willd. ex Koltz.). The trial was conducted in seven greenhouses in Methuen, Massachusetts from August through December 1997 and employed commercial poinsettia production practices. Two whitefly species, *Trialeurodes vaporariorum* (Westwood) and *Bemisia argentifolii* Bellows and Perring (= *Bemisia tabaci* [Gennadius] strain B), were present. Three treatments were examined: (1) *E. eremicus* used alone at a release rate of three females per plant per week (two greenhouses); (2) *E. eremicus* at an intermediate release rate of two females per plant per week, combined with mid-season use of buprofezin (two applications, spaced 1 week apart, applied in weeks 9 and 10) (two greenhouses); and (3) *E. eremicus* at a low release rate of one female per plant per week, combined with mid-season use of buprofezin, applied as in treatment 2 (two greenhouses). In addition, observations were made in one additional greenhouse at the site, in which the grower used pesticides for whitefly control. Prior to the start of the trial, cuttings used for all treatments experienced some pesticide use, first abamectin during rooting and later buprofezin at potting to reduce whitefly numbers, which were initially very high. At harvest, densities of live whitefly nymphs were not statistically different among the biological control treatments, indicating that a low parasitoid release rate combined with buprofezin was as effective as a higher release rate of the parasitoid used alone. Nymphal densities in separate market samples (based on smaller sample sizes) showed differences among treatments, but all treatments, including the low parasitoid release rate + buprofezin maintained densities of live nymphs + pupae at or below approximately two per leaf, a level commercially acceptable in local markets. Control costs per single-stemmed poinsettia plant were $1.18 for the high parasitoid release treatment, $0.75 for the treatment of weekly releases of two female parasitoids per plant per week + buprofezin, $0.38 for the treatment of releases of one female parasitoid per plant per week + buprofezin, and $0.14 for the chemical control greenhouse. © 2000 Academic Press

***Key Words:* *Eretmocerus eremicus; Trialeurodes vaporariorum; Bemisia argentifolii;* poinsettia; inundative biological control; efficacy; cost; insect growth regulator; integration; buprofezin; reduced risk pesticides; IPM.**

INTRODUCTION

The whiteflies *Trialeurodes vaporariorum* (Westwood) and *Bemisia argentifolii* Bellows and Perring (= *Bemisia tabaci* [Gennadius] strain B) (Homoptera: Aleyrodidae) are important pests on poinsettia (*Euphorbia pulcherrima* Willd. ex Koltz.) (Helgesen and Tauber, 1974; Byrne *et al.*, 1990). Previous research in our laboratories has assessed the relative efficacy and cost of three species or strains of parasitoids (*Encarsia formosa* Gahan, *E. formosa* Beltsville strain, and *Eretmocerus eremicus* Rose and Zolnerowich) for control of *B. argentifolii* (Hoddle and Van Driesche, 1996; Hoddle *et al.*, 1997a,b, 1998a,b). We have examined the effect of two fixed release rates and two variable release strategies (Hoddle *et al.*, 1999). Our previous trials have been run at both the scale of small research greenhouses containing 90 poinsettia plants and that of large commercial greenhouses holding 1000–3000 poinsettia plants.

1049-9644/00 $35.00
Copyright © 2000 by Academic Press
All rights of reproduction in any form reserved.

Our past trials have indicated that of the three parasitoid species or strains tested, *E. eremicus* was most effective in suppressing population growth of *B. argentifolii* (Hoddle *et al.*, 1997a,b, 1998a). For *E. eremicus*, of the two release rates tested (three females per plant per week and one female per plant per week), survivorship of whiteflies in monitored cohorts in small greenhouses was lower in greenhouses receiving the higher release rate (Hoddle *et al.*, 1998a). Numbers of live nymphs per leaf at harvest in greenhouses receiving the lower release rate were inconsistent, sometimes being lower than those in greenhouses receiving three parasitoids per plant per week (Hoddle *et al.*, 1998a), but in other trials rising rapidly to unacceptable levels after 12 weeks (J. P. Sanderson *et al.*, unpublished). Varying the release rate within the cropping season resulted in only slight or no appreciable increase in whitefly suppression (Hoddle *et al.*, 1999).

Trials in large commercial greenhouses on poinsettia stock plants over summer showed that the higher release rate of *E. eremicus* resulted in cuttings that at harvest had *B. argentifolii* nymphal densities that were acceptable to growers, but releases of *E. formosa* Beltsville strain at the same rate did not successfully control *B. argentifolii* (Hoddle and Van Driesche, 1999). Releases of *E. eremicus* at three females per plant per week, however, were not economically competitive with chemical control (imidacloprid, which provided season-long control of *B. argentifolii* on poinsettia with one application), and biological control was not affordable by producers in view of production costs and the product's wholesale value.

The cost of whitefly biological control on poinsettia could be reduced if the release rate of *E. eremicus* was lowered and the parasitoid was supplemented with mid-crop use of a compatible insect growth regulator (IGR). Previously, we have shown that four of five insect growth regulators tested caused very little mortality to either adults, older larvae, or pupae of *E. eremicus* (Hoddle *et al.*, 2000). Of these insect growth regulators, two (buprofezin and fenoxycarb) were evaluated in small greenhouses holding 90 poinsettias. One IGR, buprofezin, was found to give excellent control of *B. argentifolii* when combined with a low release rate (one female parasitoid per plant per week) of *E. eremicus* (J. P. Sanderson *et al.*, unpublished), but was less effective when used alone. Here, we report the results of a trial in a commercial poinsettia crop infested with mixed whitefly species (*T. vaporariorum* and *B. argentifolii*) in which the relative effectiveness and costs of the use of a high release rate of *E. eremicus* alone were compared to those of lower parasitoid release rates (either two or one female per plant per week) that were combined with applications of the insect growth regulator buprofezin.

MATERIALS AND METHODS

Overall Experimental Design

This experiment was conducted in 1997 using seven greenhouses owned by Loosigian Farms, Inc. in Methuen, Massachusetts. Three treatments were examined: (1) *E. eremicus* used alone at a release rate of three females per plant per week; (2) releases of *E. eremicus* at an intermediate rate of two females per plant per week, combined with mid-season use of buprofezin (two applications, spaced 1 week apart, applied in weeks 9 and 10 of the trial); and (3) *E. eremicus* released at a low rate of one female per plant per week, combined with mid-season use of buprofezin, applied as in treatment 2. Each of these three treatments was replicated twice, each in a separate greenhouse. In addition, whitefly densities were monitored in a seventh greenhouse at the same site in which the grower controlled whiteflies with applications of conventional pesticides. In all six greenhouses of treatments 1 to 3, two control cages were stocked with plants from the greenhouse in which they were set up, and whitefly numbers were monitored on the caged poinsettias. One cage in each house was designated as the control and received no suppressive treatment for whitefly population growth. The second cage was a control for the effect of caging on whitefly population growth and was treated with parasitoids and buprofezin in the same manner as the greenhouse in which the cage was located.

Greenhouse Characteristics, Cultivars, and Plant Management

All of the experimental greenhouses were 50 m long and covered with a single layer of plastic. All greenhouses were 5.5 m wide except one, used as one replicate of treatment 3 (low parasitoid release rate + buprofezin), which was 7.3 m wide. The five, same-sized greenhouses had an average of 1152 (range 1020–1250) poinsettia plants, of various cultivars (Table 1). Most plants were single-stem plants in 16.5-cm-diameter pots, but larger pots with double- or triple-stemmed plants were also present in some greenhouses (see Table 1 for details). The one larger greenhouse (replicate 2 of treatment 3) contained 2340 plants. The chemical control greenhouse contained 800 plants. Unrooted cuttings were purchased from several suppliers (Paul Ecke Ranch, Encinitas, CA; Yoder Brothers, Barberton, OH; and Fischer USA, Inc., Boulder, CO) and rooted on site by the grower. Because cuttings were subject to on-site infestation by greenhouse whitefly from adjacent areas used for retail sale of foliage plants, all cuttings were treated in the rooting room with diflubenzuron (Adept; Uniroyal Chem. Co., Middlebury, CT) for whitefly on 13 August. Potting of poinsettia cuttings occurred approximately 5 days af-

ter the diflubenzuron application and greenhouse benches were immediately filled with pots spaced at final planting density. Densities of whitefly nymphs on cuttings in the week of potting were determined on 18 and 19 August by examination of all leaves on 50 cuttings in each greenhouse. Densities ranged from 0.5 to 2.0 nymphs per leaf (mean 1.2 ± 0.2 [SE]). Initial densities on cuttings were too high for commencement of a biological control program with *E. eremicus.* Consequently, all plants in the entire trial (all greenhouses in the three treatments, the chemical control greenhouse, and all plants in all control cages) were treated with buprofezin (Accolade, AgroEvo USA Co., Wilmington, DE), at 0.27 g a.i. per L water, applied to runoff on 20 August to lower whitefly nymphal densities to levels (range 0.00–0.25, mean 0.08 nymphs per leaf) that we had observed previously on rooted cuttings obtained from commercial sources (Hoddle *et al.*, 1997a,b, 1998a, 1999). During the trial, all plants were managed in the same manner (e.g., fertilization, watering, micronutrient management, and fungus gnat control), except for whitefly control practices.

Cage Construction and Filling with Plants

Control cages were constructed of PVC pipe frames (153 × 92 × 117 cm) covered with fine mesh screening with a 95-μm opening size. Each cage had two sleeves through which plants could be handled and one clear vinyl window between sleeves through which plants could be examined for whiteflies. Twelve plants from the population of poinsettias in each greenhouse were placed in each of the two cages in a greenhouse. These plants were selected from the original 50 examined so that the average per-leaf whitefly nymphal density on the plants chosen equaled that of the greenhouse, as measured in the initial count of whiteflies on cuttings made at the start of the trial.

Determining Relative Proportions of Whitefly Species

Because both *B. argentifolii* and *T. vaporariorum* occurred in each greenhouse, we determined the relative abundance of these two species on four occasions (18–26 August, 1–9 October, 4–11 November, and 2–4 December) by examining whitefly pupae and fourth instar parasitized nymphs. We also examined exuviae of fourth instar nymphs from which parasitoids had successfully emerged to determine whether parasitoids were more successful at emerging from one whitefly species or the other. These observations were made in the greenhouses by examining individuals of these stages that were encountered during the observations made to estimate whitefly densities with both a head-mounted magnifier ("Optivisor," Donegan Optical Company, Inc., Lenexa, KS) and a hand lens (10–16×) to provide additional magnification. Numbers of stages examined on the four sampling occasions to determine whitefly species were 219, 1314, 1339, and 743, respectively. Numbers of exuviae examined from which parasitoids had emerged were 0, 98, 1298, and 991. To determine whether either whitefly species was parasitized at a higher rate, the numbers of parasitized fourth instar nymphs of each species were compared by a χ^2 test to the numbers of unparasitized whitefly pupae of these species in the same sampling periods.

Pesticide Treatments

In each greenhouse in which buprofezin (Accolade) was a constituent of the treatment, this insect growth regulator was applied at 0.27 g a.i. per L water. In greenhouses with the highest levels of whiteflies at the end of the experiment, applications of sulfotep (Fulex Dithio; Fuller Systems, Inc., Woburn, MA) were made to reduce numbers of adult whiteflies before harvest and sale. Copies of the grower's pesticide records were obtained for the chemical control greenhouse and used to compute the number of pesticide applications and their cost, for comparison to costs of treatments applied in test greenhouses.

Parasitoid Release Rates

In each week of this study, approximately 52,000 *T. vaporariorum* fourth instar nymphs parasitized by *E. eremicus* (not mixed with sawdust) were received from Koppert Biological Systems (Berkel en Rodenrijs, The Netherlands, through the North American office in Romulus, MI). From each shipment, the number of parasitized whitefly nymphs in each of 10 20-mg subsamples was counted under a dissecting microscope at 25× magnification and the mean (± SE) of these values was used to calculate the mean number of parasitized whitefly nymphs per gram of product. This value was multiplied by the running mean (from all earlier weeks of the trial) of parasitoid sex ratio (proportion of parasitoids emerging in samples held in the laboratory that were female) and the running mean of the parasitoid emergence under greenhouse conditions (proportion of fourth instar whitefly nymphs placed in test greenhouses from which parasitoids emerged). The resulting value (estimate of nymphs per gram × parasitoid sex ratio × % parasitoid emergence) was used to determine the weight of parasitized nymphs to be distributed in each greenhouse each week.

Parasitized fourth instar nymphs used to make parasitoid releases were placed in test greenhouses weekly in 15 evenly dispersed styrofoam release cups (6 cm tall, 5.5 cm wide at bottom, 8.5 cm wide at top) glued to wooden stakes (50 cm long) at a height of 25 cm above the top of the plants. The cups had screened bottoms covered with fine meshed organdy, to allow drainage should cups get wet from overhead watering. In each greenhouse that received parasitoids, there were two sets of release cups (15 in each set), of which one set

TABLE 1

Poinsettia Cultivars, Pot Number and Size, and Plant Number for Greenhouses in Various Experimental Treatments at Loosigian Farms, Methuen, Massachusetts, in 1997

Treatment	Cultivars	Pot diameter (cm)	Plant No.
3 par., rep. 1	Bonita Red and Cortez Red	16.5 (1234 pots)	1234
3 par., rep. 2	Bonita Red and Cortez Red	16.5 (1131 pots)	1131
2 par. + IGR, rep. 1	Peterstar Red	16.5 (408 pots), 17.8 (232 pots), 20.3 (126 pots)	1250
2 par. + IGR, rep. 2	Bonita Red and Cortez Red	16.5 (1020 pots)	1020
1 par. + IGR, rep. 1	Sonora White, Sonora Pink, Jingle Bells, and Cortez Pink	16.5 (1126 pots)	1126
1 par. + IGR, rep. 2	Sonora Pink, Marble, Sonora White, Bonita Red, Cortez Red, and Jingle Bells	17.8 (1170 pots)	2340
Chemical control	Marble, Angelica, V-17, and Pink Peppermint	16.5 (500 pots), 20.3 (100 pots)	800

was labeled "A" and the other "B." In week 1 of the trial, parasitized whitefly nymphs were placed in the "A" release cups. In week 2, new parasitized whitefly nymphs were placed in the "B" set of release cups. In week 3, samples of exuviae were collected from each of the "A" release cups (which at the time of collection had been in the greenhouses for 2 weeks) to estimate emergence. Remaining exuviae were removed and then new parasitized whitefly nymphs were placed in these cups and left for parasitoids to emerge. The same pattern was repeated weekly. Allowing parasitoid pupae to remain in cups for 2 weeks before retrieving remains to estimate the rate of parasitoid emergence allowed sufficient time for all live parasitoids to emerge.

To enable expression of parasitoid release rates as either female numbers per plant or numbers per leaf, the number of mature leaves on randomly selected plants was recorded in each greenhouse on 18 August (50 plants), 11 September (30 plants), 23 October (30 plants), and 2 December (30 plants) to estimate mean leaf density per plant over the growing season.

Parasitoid sex ratio. To estimate the sex ratio of parasitoids emerging from material used in this trial, 300 parasitized whitefly nymphs from each week's shipment were held in petri dishes in a growth chamber at 22°C and long day-light regime (16L:8D h). One week later, samples were frozen. To determine sex ratio, 100 randomly selected adult parasitoids were examined under a dissecting microscope at 50× magnification and numbers of females and males were recorded.

Parasitoid emergence rates. To determine the proportion of parasitized whitefly nymphs from which adult *E. eremicus* successfully emerged while in experimental greenhouses, exuviae (which included whitefly nymphs with parasitoid exit holes and whitefly nymphs that had died) were collected from test greenhouses, taken to the laboratory, and examined under a microscope at 25×. One hundred and fifty exuviae were chosen at random from each greenhouse and examined. Those with round dorsal holes (characteristic of *E. eremicus* emergence) were counted, as were those without emergence holes (dead individuals). The mean (± SE) percentage of emergence for each greenhouse was calculated as (No. nymphs with parasitoid exit holes/No. nymphs with parasitoid exit holes + No. dead whitefly nymphs) × (100). The weekly emergence rate for the experiment as a whole was calculated by pooling the samples for all six greenhouses in which parasitoids were released into one sample of 900 whitefly nymphs.

TABLE 2

Percentage of Whitefly Stages (in a Mixed *T. vaporariorummed/B. argentifolii* Population) That Were *T. vaporariorum*, Based on Samples Summed across All Biological Control Greenhouses at Loosigian Farms, Methuen, Massachusetts, in 1997

	% *T. vaporariorum*		
Sample date	Of live 4th instar nymphs (n = sample size)	Of visibly parasitized whiteflies (n = sample size)	Of whiteflies yielding parasitoids (n = sample size)
18 to 26 Aug.	81.7 (219)	—	—
9 Sept. to 9 Oct.	80.5 (1314)	84.5 (342)	91.8 (98)
6 to 13 Nov.	84.2 (1339)	88.9 (806)	92.5 (1298)
11 Nov. to 2 Dec.	81.0 (743)	91.2 (662)	89.3 (991)
Totals	82.1 (3615)	88.9 (1810)	91.1 (2387)

Note. All whiteflies in samples that were not *T. vaporariorum* were *B. argentifolii.*

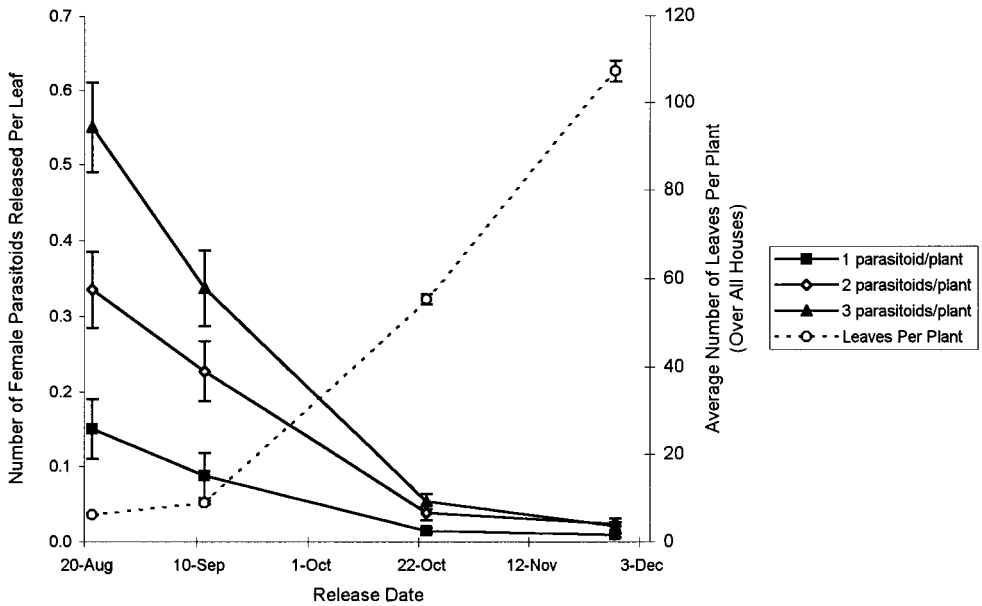

FIG. 1. Release rates of female *Eretmocerus eremicus* expressed as numbers per leaf rather than per plant to illustrate the dilution of parasitoids as foraging area to be searched increases due to plant growth over the course of the poinsettia crop.

Monitoring Whitefly Population Densities and Parasitism

To estimate whitefly population densities, three leaves (one bottom, one middle, and one top) on 90 plants in each greenhouse (n = 270) were inspected weekly for whiteflies. In the control and parasitoid release cages, three leaves on each of eight plants per cage (n = 24) were examined weekly. The number of nymphs, dead nymphs, pupae, dead pupae, parasitized fourth instar nymphs, exuviae from emerged white-

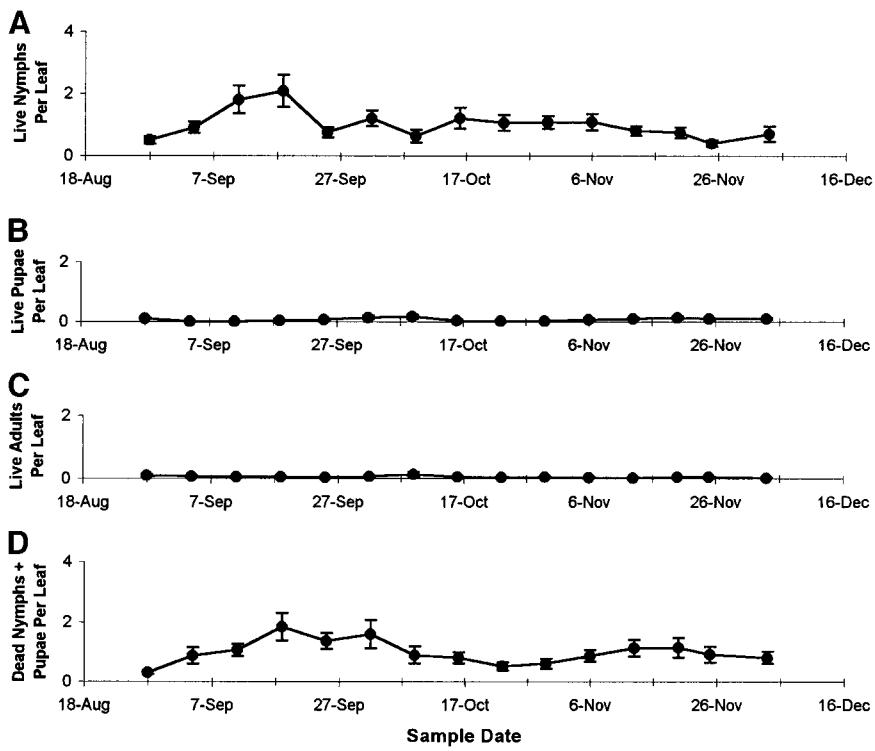

FIG. 2. Numbers per leaf of whitefly stages in a biological control greenhouse receiving three *Eretmocerus eremicus* females per plant per week (replicate 1), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D).

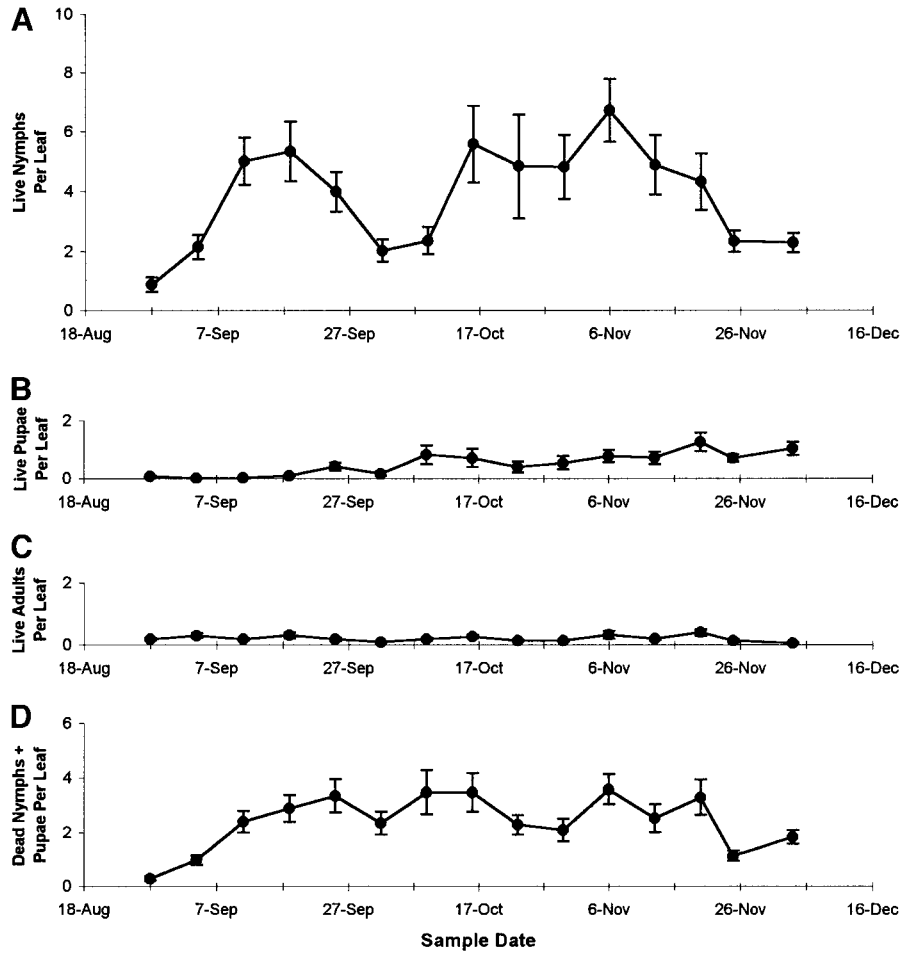

FIG. 3. Numbers per leaf of whitefly stages in a biological control greenhouse receiving three *Eretmocerus eremicus* females per plant per week (replicate 2), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D).

flies, exuviae of parasitized whiteflies with parasitoid emergence holes, and adult whiteflies were recorded. The seasonal parasitism rate in each greenhouse was calculated by pooling data from all sampling dates over the experiment within each greenhouse. Using these pooled data, the parasitism rate was determined by dividing the number of visibly parasitized whitefly nymphs by all counted fourth instar nymphs + pupae and parasitized whitefly nymphs.

End of Crop Sales Inspection

At week 16 of the trial (time of harvest), six leaves (two leaves from the bottom, two from the middle, and two from the top) on 15 randomly selected plants in each greenhouse were examined, and live nymphs, pupae, and adults were counted. These samples were collected in this manner so as to exactly match the sampling approach used in market surveys in Massachusetts and New York. Whitefly densities in the greenhouses in our trial, collected in this manner, were then compared to those on plants examined at 20 retail outlets in Massachusetts in December 1997.

RESULTS

Whitefly Species Composition

A mean of 82.1% of the live, not visibly parasitized fourth instar whitefly nymphs in the test greenhouses were *T. vaporariorum* and the remainder were *B. argentifolii.* There was no obvious change over time in the percentage of such nymphs that were *T. vaporariorum* (range 80.5–84.2%) (Table 2). Of the visibly parasitized whitefly nymphs, more were *T. vaporariorum* (88.9%). This difference between the percentage of not-visibly parasitized nymphs that were *T. vaporariorum* and the percentage of visibly parasitized nymphs that were this species was statistically significant ($\chi^2 = 42$, $df = 1$, $P < 0.05$). Of parasitized whiteflies from which parasitoids had successfully emerged (seen in samples as exuviae), the percentage that was *T. vaporariorum* was still higher, 91.1%. The percentage of individuals in this category (successful parasitoid emergence) that were *T. vaporariorum* (91.1%) was significantly different from the percentage of visibly parasitized live nymphs that were *T. vaporariorum* (88.9%), although

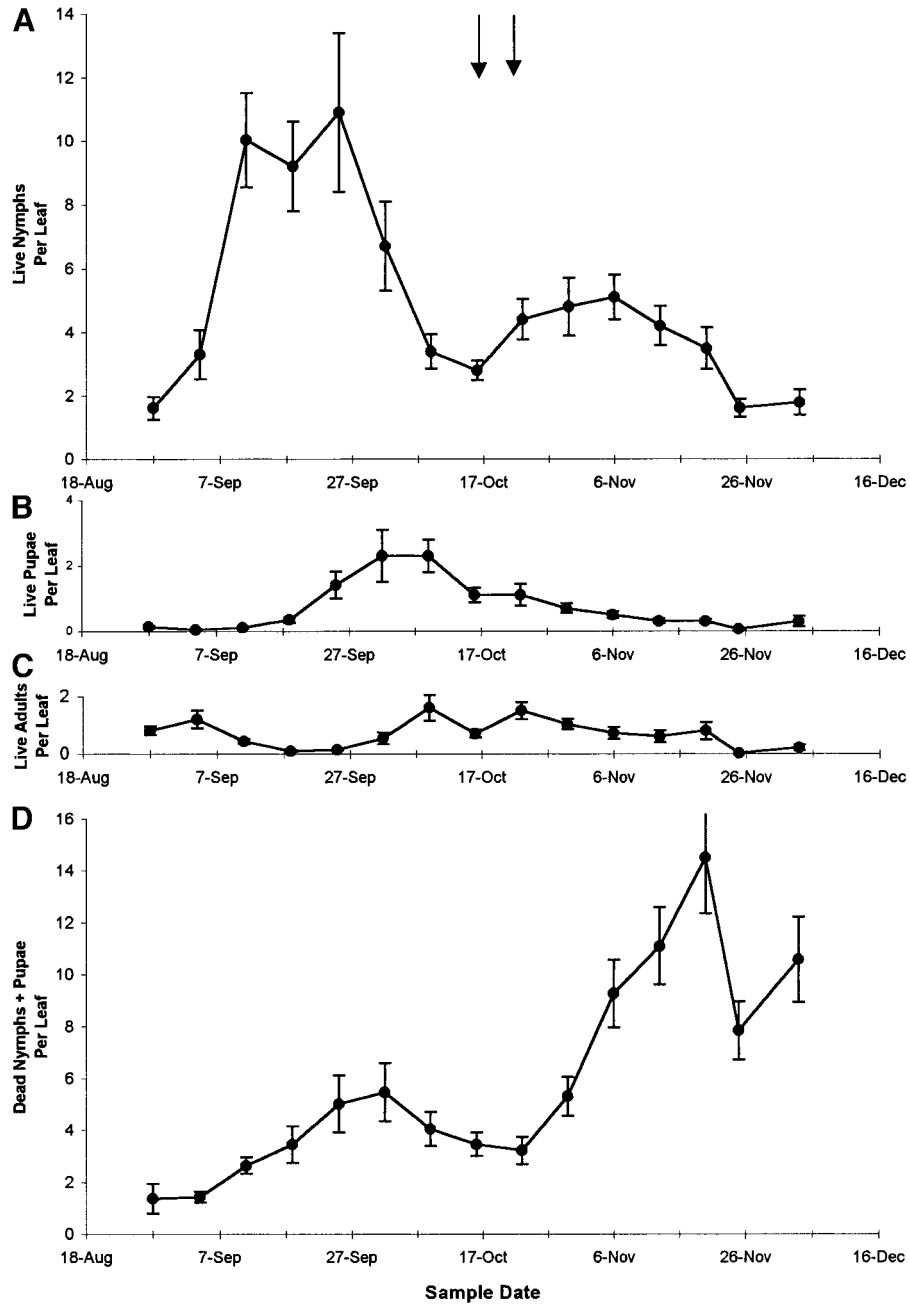

FIG. 4. Numbers per leaf of whitefly stages in a biological control greenhouse receiving two *Eretmocerus eremicus* females per plant per week plus two mid-season applications of buprofezin (replicate 1), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D). (Arrows indicate dates of buprofezin applications.)

this difference was rather small ($\chi^2 = 5.0$, $df = 1$, $P < 0.05$).

Parasitoid Release Rates Achieved

The grand mean (± SE) of the percentage of emerging parasitoids that were female in samples held in the laboratory from shipments received between 21 August and 2 December was 45.2 ± 1.0%. The grand mean (± SE), across all dates and greenhouses, of the percentage of parasitoid pupae from which adults emerged while in test greenhouses was 64.3 ± 3.9%. Season-long mean emergence rates among test greenhouses varied from 61% ± 3 to 69 ± 1%, and among dates within greenhouses, the variation was greater, from 17 ± 3 to 91 ± 3%.

Season-long average release rates and SE values (as female wasps per plant per week) in six test greenhouses were as follows, relative to their intended treat-

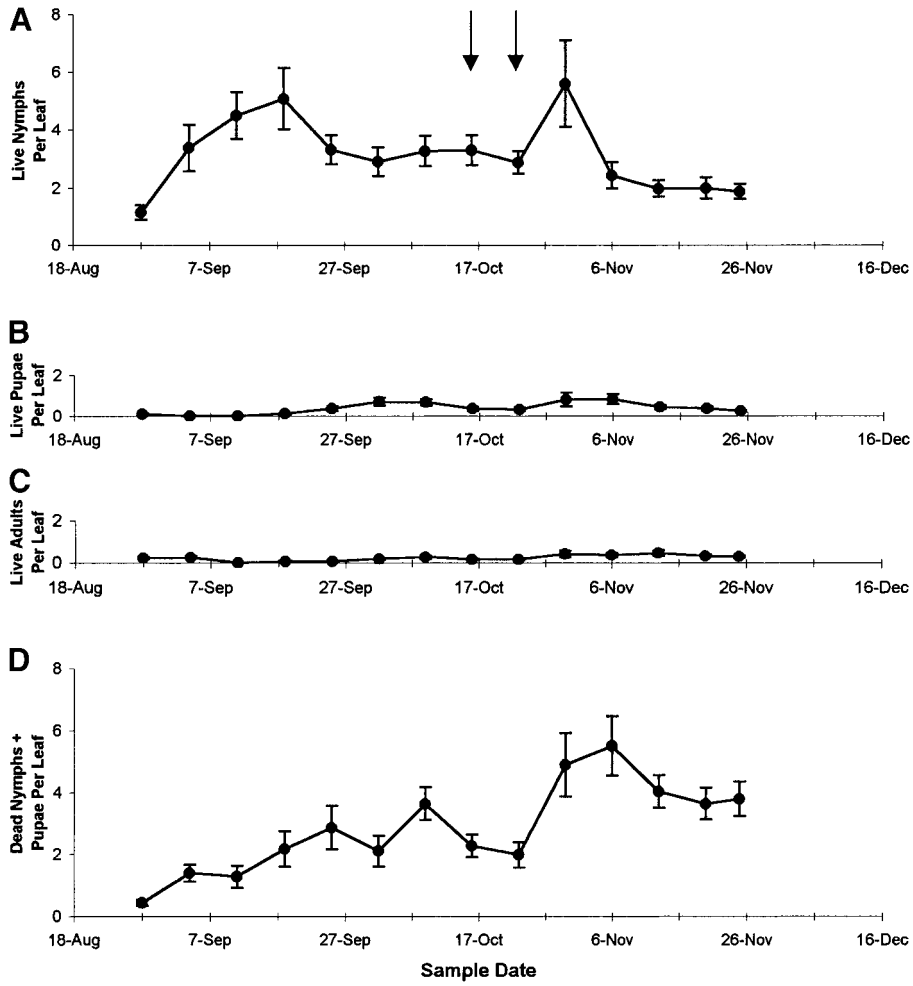

FIG. 5. Numbers per leaf of whitefly stages in a biological control greenhouse receiving two *Eretmocerus eremicus* females per plant per week plus two mid-season applications of buprofezin (replicate 2), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D). (Arrows indicate dates of buprofezin applications.)

ments: intended rate of 3.0, achieved rates of 2.5 ± 0.2 and 2.5 ± 0.3; intended rate of 2.0, achieved rates of 1.9 ± 0.1 and 1.8 ± 0.1; and intended rate of 1.0, achieved rates of 0.9 ± 0.1 and 0.9 ± 0.1. Numbers of leaves per plant increased a mean of 17.3-fold (from a mean across the six test greenhouse of 6.2 [0.1 ± SE] leaves on 18 August to 107.2 [2.4 ± SE] leaves on 2 December), causing the parasitoid release rate per leaf to drop proportionally over the course of the trial (Fig. 1).

Whitefly Population Densities

Trends in densities per leaf of whitefly nymphs, pupae, adults, and dead immature whiteflies seen in population monitoring during the trial are given in Figs. 2–8. There were no large differences in population densities among treatments. After reaching maximum values of 2–11 live nymphs per leaf in the middle of the growing season (mid-September to the end of October), whitefly populations in all greenhouses declined. Final densities (± SE) at harvest of live whitefly nymphs per leaf, averaged across the two greenhouses for each treatment, were 1.49 (± 0.20) for the release of three female parasitoids per plant per week without IGR application, 1.83 (± 0.24) for the release of two females plus IGR application, and 1.41 (± 0.23) for the release of one female plus IGR application (see Table 3A). The three biological control treatments differed significantly in final whitefly nymphal density from that in the chemical control greenhouse (0.28 ± 0.08 SE) (ANOVA, $F = 8.63$, $df = 3$, $P = 0.0001$), but all treatments produced plants that were acceptable for the market, being at or below the threshold of approximately 2 live nymphs + pupae per leaf (with live nymph + pupae densities per leaf for the high, medium and low parasitoid release rate treatments being 2.05 ± 0.23 [SE], 2.09 ± 0.25 [SE], and 1.57 ± 0.24 [SE], respectively). More importantly, in relation to the purpose of this trial, we found that the three biological control treatments did not differ

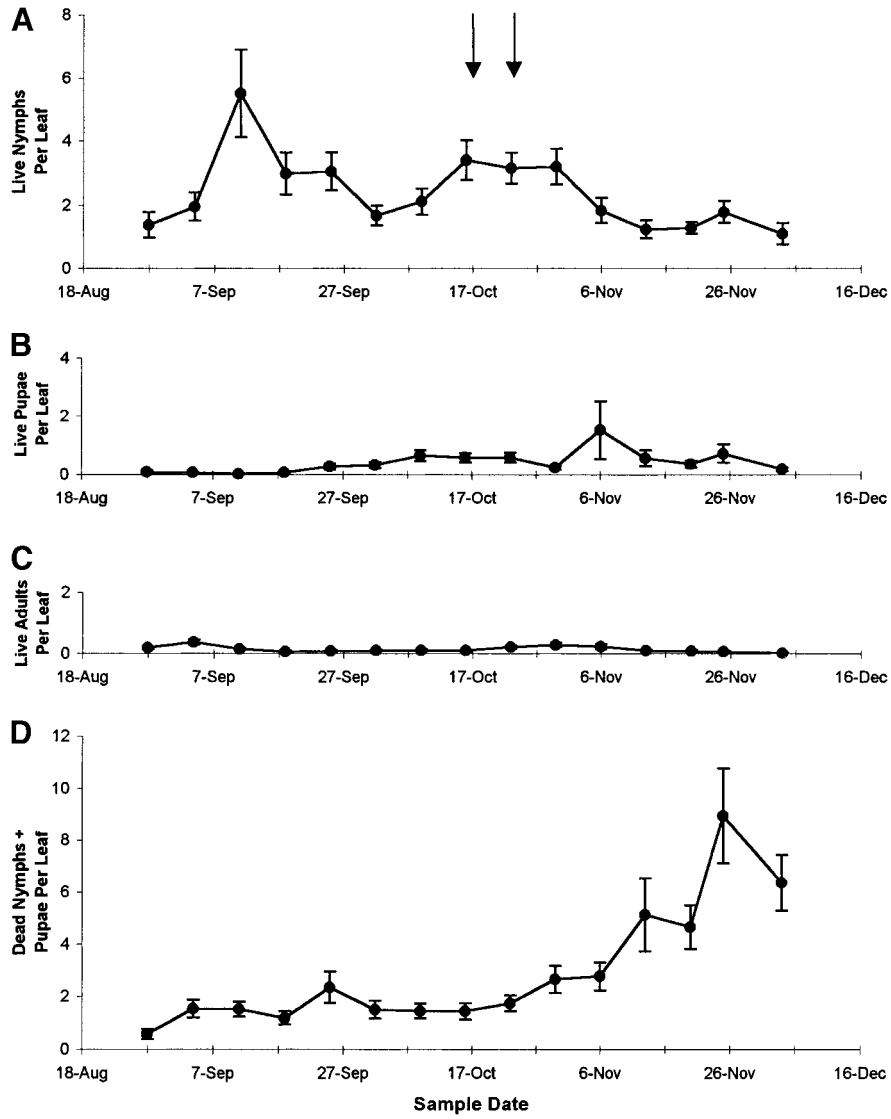

FIG. 6. Numbers per leaf of whitefly stages in a biological control greenhouse receiving one *Eretmocerus eremicus* female per plant per week plus two mid-season applications of buprofezin (replicate 1), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D). (Arrows indicate dates of buprofezin applications.)

significantly among themselves in terms of final whitefly nymphal density, indicating that reduced parasitoid release rates were as effective as the high rate when lower rates were combined with applications of the IGR buprofezin.

Whitefly Densities in Cages

In all six greenhouses, nymphal whiteflies in control cages (receiving no whitefly suppression measures) reached higher seasonal peak densities than did whiteflies in treatment cages in the same greenhouse (i.e., cages receiving the same treatment as the particular greenhouse); however, in one replicate of the highest parasitoid release rate, the difference was trivial (Table 4). Seasonal peak densities in control cages ranged from 1.0 to 38.0 live nymphs per leaf and those in treatment cages ranged from 0.8 to 4.8 live nymphs per leaf.

To calculate the degree to which parasitoids suppressed whitefly densities inside cages, we compared the seasonal maximum values in control and treatment cages (Table 4). For each greenhouse, the seasonal maximum density of live whitefly nymphs was divided by that in the treatment cage and then the values for the two greenhouses were averaged. In cages, the high parasitoid release rate (three female parasitoids, no IGR) reduced nymphal densities on average to 44.0% of control densities, the intermediate parasitoid release rate (two female parasitoids plus IGR) reduced nymphs to 30.5% of control densities, and the low parasitoid release rate (one fe-

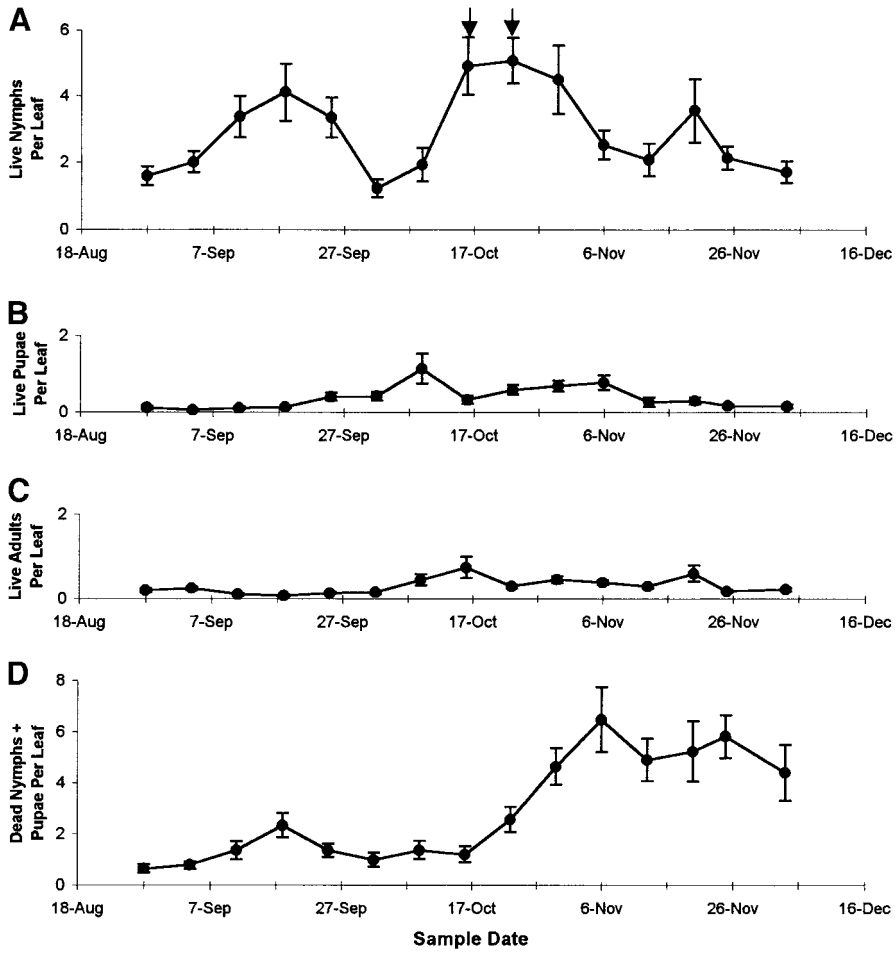

FIG. 7. Numbers per leaf of whitefly stages in a biological control greenhouse receiving one *Eretmocerus eremicus* female per plant per week plus two mid-season applications of buprofezin (replicate 2), where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D). (Arrows indicate dates of buprofezin applications.)

male parasitoid plus IGR) reduced nymphs to 25.8% of control densities.

Parasitism Rates in Test Greenhouses

Parasitism in the two greenhouses receiving the highest parasitoid release rate (pooled across the two replicate greenhouses) peaked in mid-to-late October at 43.8% (± 7.0% for 95% C.I., $n = 194$) and then declined (Figs. 9E and 9F). In greenhouses receiving lower parasitoid release rates plus IGR applications, parasitism rates either reached high levels later or remained high longer (see Figs. 9E and 9F versus 9A–9D) than in greenhouses with the high parasitoid release rate. Peak parasitism levels (pooled across the two replicate greenhouses for each treatment) were 40.5% (± 6.4% for 95% C.I., $n = 227$) where two female parasitoids were released per plant per week and 69.9% (± 5.0% for 95% C.I., $n = 316$) where one female parasitoid was released per plant per week (Figs. 9A–9D). Some parasitism by *E. eremicus* occurred in the chemical control greenhouse because of movement of parasitoids among greenhouses, which were physically separate, but adjacent. Parasitism in the chemical control greenhouse peaked at 42.0% (± 9.7% for 95% C.I., $n = 100$) on 2 October, but declined to zero by 30 October and remained zero thereafter.

The rates of parasitism, pooled over all dates for both replicates of a given treatment, were 23.9% (± 1.5% for 95% C.I., $n = 3091$) where three female parasitoids per plant per week were released, 25.8% (± 1.1% for 95% C.I., $n = 5871$) where two female parasitoids were released in combination with buprofezin, and 29.4% (± 1.3% for 95% C.I., $n = 4508$) where one parasitoid female was released in combination with buprofezin (see Table 5 for seasonal percentage parasitism values for each greenhouse). These values indicate that buprofezin's use was not detrimental to parasitism and that lowering the release rate does not cause the parasitism rate to decrease; rather it may be associated with small increases in the parasitism rate.

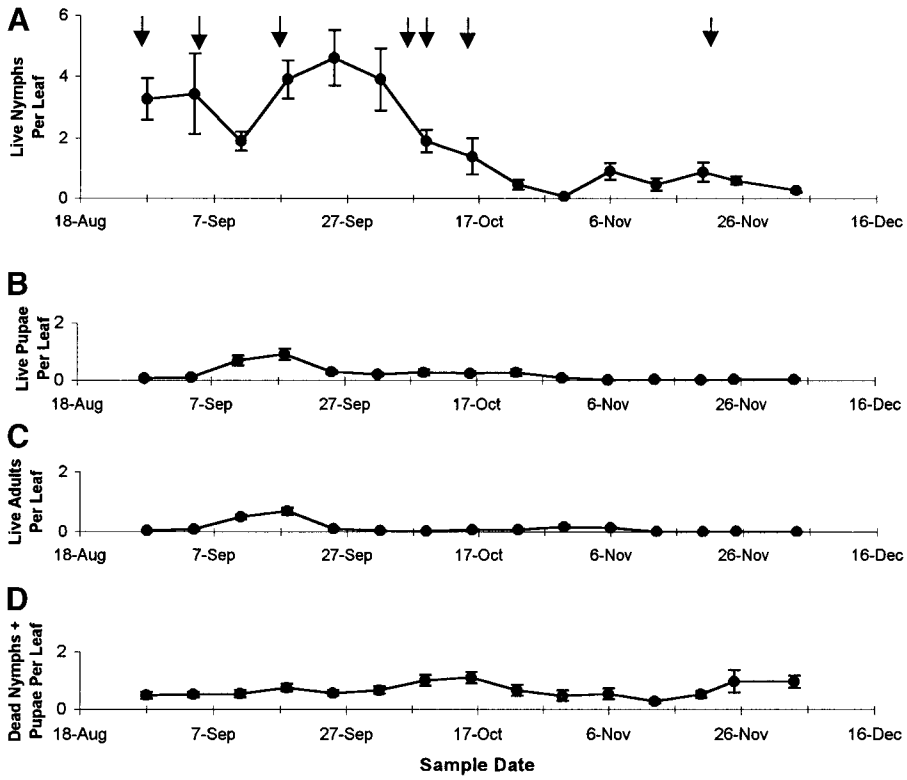

FIG. 8. Numbers per leaf of whitefly stages in a chemical control greenhouse, where stages counted were live nymphs (A), pupae (B), adults (C), and dead nymphs + pupae (D). (Arrows indicate dates of applications for whitefly control.)

Pesticide Applications in Chemical Control Greenhouse

Seven pesticide applications were made, using label rates, in the chemical control greenhouse to suppress whitefly populations: 28 August and 4 September (abamectin, Avid; Novarits), 17 September (imidacloprid, Marathon; Olympic Horticultural Products, Mainland, PA), 4 October (orthene, PT 1300; Whitmire Micro-Gen. Res. Labs), 17 October (buprofezin, Accolade; AgroEvo), and 8 and 21 November (sulfotep, Fulex Dithio; Fuller Systems, Inc.).

End of Crop Plant Quality and Treatment Costs

At harvest, densities of whitefly nymphs in the separate market sample differed between the lowest para-

TABLE 3

Comparison of Final Whitefly Densities (Numbers per Leaf, Mean and SE) in Test Greenhouses in Trial, Using Two Sampling Approaches (Research Population Counts-A; Harvest Market Survey Method-B), to Densities (1) in the Chemical Control Greenhouse and (2) at 20 Retail Outlets Offering Poinsettia for Sale in Massachusetts

Whitefly life stage	High release, no IGR	Intermediate release + IGR	Low release + IGR	Chemical control	Retail outlets
		A. Numbers among greenhouses in trial (research population counts)[a]			
Nymphs	1.49 ± 0.20A	1.83 ± 0.24A	1.41 ± 0.23A	0.28 ± 0.08B	—
		B. Numbers using harvest market survey method[b]			
Nymphs	1.11 ± 0.27A	0.85 ± 0.18AC	1.93 ± 0.38B	—	0.41 ± 0.08C
Pupae	0.36 ± 0.14A	0.07 ± 0.02A	0.14 ± 0.04A	—	0.25 ± 0.13A
Adults	0.03 ± 0.01A	0.02 ± 0.01A	0.07 ± 0.03A	—	0.06 ± 0.02A

[a,b] Research population counts were taken by examining 3 leaves (one top, middle, and bottom, each) from 90 plants per greenhouse per date. Counts from harvest market survey method were taken by examining 6 leaves (two top, middle, and bottom, each) from 15 plants; counts in research greenhouses were taken by this second method in addition to more intensive research population counts to allow for direct comparison to values in retail outlet survey. Values in the same row followed by the same letter did not differ significantly in a Tukey's Studentized range test.

TABLE 4

Seasonal Maximum Values for Mean Numbers of Whitefly Stages per Leaf in Control Cages and Treatment Cages in Respective Experimental Greenhouses

	Nymphs (mean ± SE)		Pupae (mean ± SE)		Adults (mean ± SE)	
Treatment/replicate	Treatment	Control	Treatment	Control	Treatment	Control
3 par., rep. 1	0.8 ± 0.3	1.0 ± 1.0	0.1 ± 0.1	0.2 ± 0.1	0.1 ± 0.01	0.04 ± 0.04
3 par., rep. 2	0.5 ± 0.5	6.3 ± 4.1	0.04 ± 0.04	1.2 ± 0.8	0.04 ± 0.04	1.3 ± 0.3
2 par. + IGR, rep. 1	4.8 ± 1.0	8.3 ± 2.3	0.6 ± 0.5	1.9 ± 1.2	0.4 ± 0.1	1.0 ± 0.3
2 par. + IGR, rep. 2	1.2 ± 0.8	38.0 ± 6.2	0.4 ± 0.3	3.7 ± 1.1	0.1 ± 0.1	5.2 ± 1.6
1 par. + IGR, rep. 1	1.0 ± 0.7	4.4 ± 2.7	0.04 ± 0.04	0.5 ± 0.3	0.1 ± 0.1	0.4 ± 0.2
1 par. + IGR, rep. 2	4.0 ± 1.7	13.8 ± 5.9	0.3 ± 0.1	1.3 ± 0.9	0.2 ± 0.2	2.3 ± 1.1

sitoid application rate and the other two higher rates ($F = 4.56$, $df = 3$, $P < 0.05$) (Table 3B), with the mean live nymphal densities being 1.93 (± 0.38) for the low parasitoid release rate plus IGR, 1.11 (± 0.27) for the high parasitoid release rate, and 0.85 (± 0.18) for the intermediate rate plus IGR. Nymphal densities for the high parasitoid + IGR and low parasitoid + IGR treatments were higher than those from plants sampled at retail outlets and this difference was statistically significant (Table 3B). No differences were found among treatments for densities of pupae ($F = 0.35$, $df = 3$, $P > 0.05$) or adults ($F = 0.68$, $df = 3$, $P > 0.05$).

Costs of treatments were calculated using a 1997 parasitoid price of $11.00 per 1000 parasitized *T. vaporariorum* nymphs (a price available if ordering in quantities of 15,000 or more pupae in bottles, rather than glued to cards, from Koppert Biological Systems) and, for buprofezin, an anticipated price supplied by The Scotts Company, which plans to market a greenhouse-labeled buprofezin product. Cost calculations for parasitoids assumed 50% female, 60% emergence, and 142% overfilling by Koppert (their action to compensate for the failure of some parasitoids to emerge), and, for buprofezin, application at the high labeled rate (see Materials and Methods). In this experiment, average treatment costs per single-stemmed poinsettia plant were $1.18 for the high parasitoid release treatment, $0.75 for the treatment of two female parasitoids + buprofezin, $0.38 for the treatment of one female parasitoid + buprofezin, and $0.14 for the chemical control greenhouse (Table 6).

DISCUSSION

In earlier laboratory work, we found buprofezin to be among the least toxic of five insect growth regulators tested against both adult *E. eremicus* and older larvae and pupae inside hosts (Hoddle *et al.*, 2000). In that earlier study, few harmful effects were observed when adult parasitoids were exposed to greenhouse-aged buprofezin residues in the laboratory. For immature stages, it was noticed that eggs and young parasitoid larvae inside living hosts on treated leaves were more susceptible than older larvae or parasitoid pupae to buprofezin (Hoddle *et al.*, 2000; see also Gerling and Sinai, 1994). Similarly, Jones *et al.* (1998) found that buprofezin had relatively little effect on adult parasitoid longevity of the related species *Eretmocerus tejanus* Rose and Zolnerowich, but was detrimental to survival of immature parasitoids whose hosts were treated with the IGR if parasitoids were exposed as first instars (5 days after oviposition). These data suggest that limited numbers of applications of buprofezin are compatible with inundative releases of *Eretmocerus* spp. as parasitoids are robust to detrimental effects.

Subsequent to our laboratory study (Hoddle *et al.*, 2000), we ran trials in small greenhouses to compare the efficacy of *E. eremicus* releases plus an IGR (buprofezin or fenoxycarb) to that of either (1) a higher rate of the parasitoid used alone, (2) the low rate of the parasitoid used alone, or (3) either IGR used alone (J. P. Sanderson *et al.*, unpublished). Buprofezin was selected because in the laboratory it had the fewest effects on *E. eremicus* of the five IGRs tested by Hoddle *et al.* (2000). For a second material to test in the small greenhouse trial, we wanted a material registered in the United States by the EPA for use in greenhouses. Only two materials of the five that we tested (kinoprene and fenoxycarb) had EPA registrations when the small greenhouse trials were run. Of these, fenoxycarb had fewer harmful effects on *E. eremicus* and for these reasons we included it in the small greenhouse trial of J. P. Sanderson *et al.* (unpublished). Results showed that a low release rate of *E. eremicus* (one female per plant per week) in combination with two mid-season applications of buprofezin was more effective than the high rate of *E. eremicus* by itself or either IGR by itself. Of the two IGRs, buprofezin provided better whitefly suppression, both when used alone and when used in combination with a low rate of parasitoid release (J. P. Sanderson *et al.*, unpublished).

Based on results from these small greenhouse trials, in this study we tested in commercial greenhouse poin-

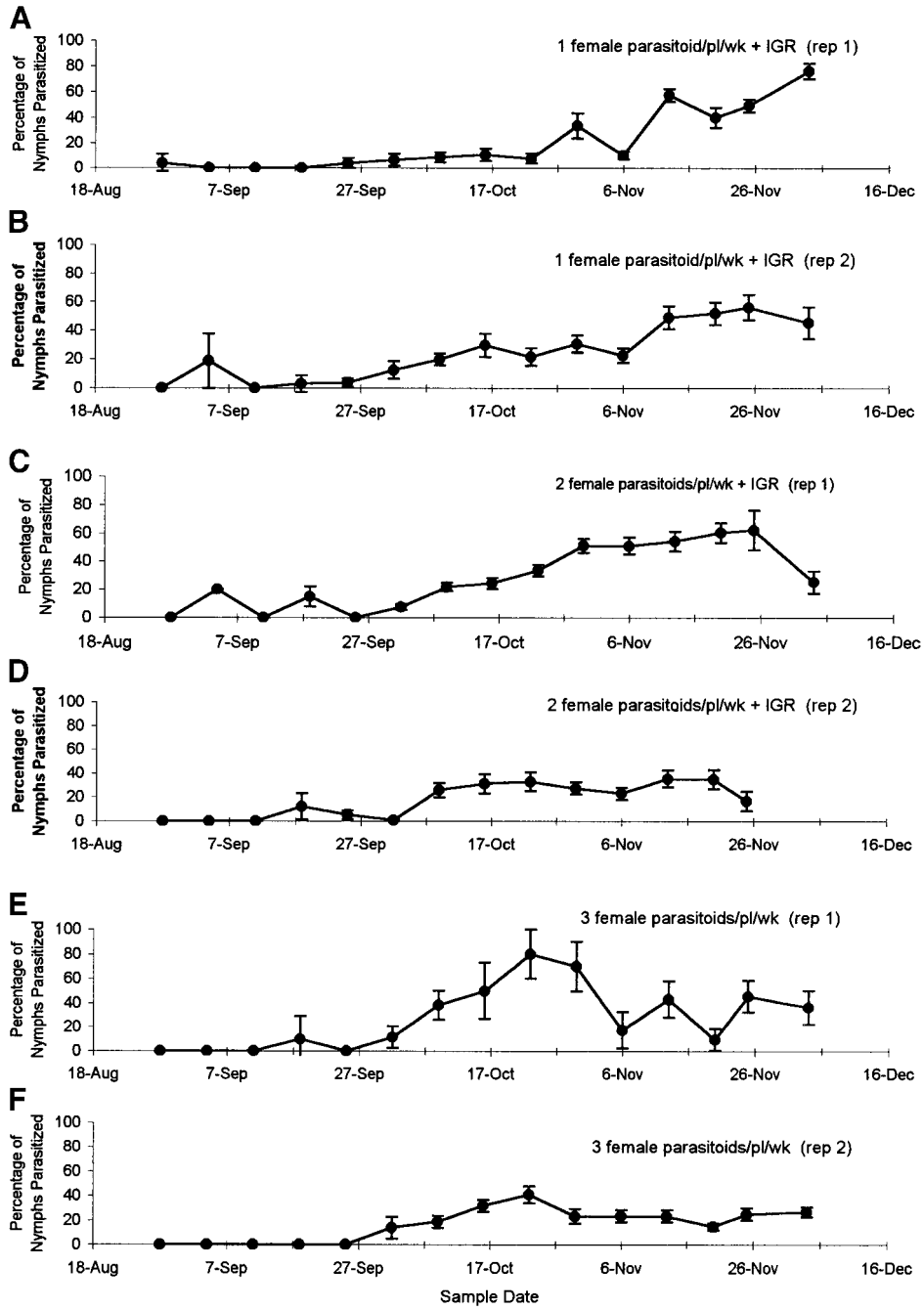

FIG. 9. Rates of parasitism (visibly parasitized nymphs/visibly parasitized nymphs plus healthy whitefly pupae) seen in population density sampling in test greenhouses over the course of the trial, for all six greenhouses.

settia crops the relative efficacy of low rates of the parasitoid *E. eremicus* when used together with buprofezin versus the efficacy of a high rate of the parasitoid used alone. We found no differences in the level of whitefly nymphs per leaf at harvest (in the density samples of the main experiment, see Table 3A) among a high rate (three females per plant per week) of *E. eremicus* used alone, an intermediate rate (two females) plus buprofezin, and a low rate (one female) plus buprofezin. In separate market samples (Table 3B), we found a statistically significant difference between the low parasitoid rate + IGR (1.93 live nymphs per leaf) and the other two parasitoid treatments (1.11 and 0.85 live nymphs per leaf for the high and intermediate release rates), but this difference was small and all treatments produced commercially acceptable plants. Also, it is of interest to note that we found no evidence for the replacement of *T. vaporariorum* by *B.*

TABLE 5

Seasonal Percentage Parasitism by *Eretmocerus eremicus* and Densities per Leaf of Whitefly Life Stages in Test Greenhouses at Time of Harvest

Treatment	Seasonal % parasitism (*n*)	Mean nymphs per leaf (SE)	Mean pupae per leaf (SE)	Mean adults per leaf (SE)
3 par., rep. 1	31.9 (426)	0.70 ± 0.25	0.10 ± 0.03	0.02 ± 0.01
3 par., rep. 2	22.6 (2665)	2.27 ± 0.32	1.03 ± 0.23	0.05 ± 0.02
2 par. + IGR, rep. 1	23.1 (1860)	1.87 ± 0.26	0.24 ± 0.05	0.29 ± 0.05
2 par. + IGR, rep. 2	27.1 (4011)	1.78 ± 0.4	0.29 ± 0.16	0.20 ± 0.10
1 par. + IGR, rep. 1	31.2 (2441)	1.10 ± 0.34	0.18 ± 0.06	0.02 ± 0.01
1 par. + IGR, rep. 2	27.3 (2067)	1.71 ± 0.32	0.15 ± 0.05	0.22 ± 0.04
Chemical control	16.2 (960)	0.28 ± 0.08	0.03 ± 0.03	0.004 ± 0.004

Note. Data are for *T. vaporariorum* and *B. argentifolii* combined.

argentifolii over the course of this study, contrary to evidence reported by Liu *et al.* (1994).

The use of buprofezin did not reduce parasitism, which was actually highest in the treatment with the lowest parasitoid release rate combined with buprofezin (Table 5). This increase in parasitism may have been due to lower host mortality due to the reduced frequency of multiple oviposition or host feeding associated with lower rates of release of this species (Hoddle *et al.*, 1998a, 1999). It is also possible that if healthy whiteflies were being killed by buprofezin but parasitized ones were not (at least ones bearing older immature parasitoids), then parasitism in samples may have been artificially raised in samples (Van Driesche, 1983). Finally, the mixture of *T. vaporariorum* and *B. argentifolii* may have promoted higher rates of parasitism by *E. eremicus* than would have occurred in the presence of *B. argentifolii* alone, given that *T. vaporariorum* is a better host for this parasitoid (Boisclair *et al.*, 1990; Szabo *et al.*, 1993).

Experimental treatments differed substantially in per-plant pest control costs, being $0.38 for one, $0.75 for two, and $1.18 for three wasps per plant per week, respectively, with buprofezin used in the first two treatments only (per-plant costs include both parasitoids and insect growth regulator applications). We speculate that further reductions of per-plant cost may be possible when using *E. eremicus* with buprofezin. Parasitoid releases were made in this experiment during the 2-week period over which buprofezin was applied. Jacobson and Chambers (1996) showed that in greenhouse tomatoes in which buprofezin was applied for control of the glasshouse leafhopper (*Hauptidia maroccana* [Melichar]), releases of *E. formosa* for whitefly control could be omitted for 10 weeks because of the protection afforded by buprofezin (in a long-season crop, with relatively low whitefly populations). We argue that the same may be likely when buprofezin is used for whitefly control on poinsettia and *E. eremicus* is the natural enemy used. Furthermore, the lowest parasitoid release rate tested (one female per plant per week) provided commercially acceptable control when combined with buprofezin. Even lower release rates of *E. eremicus* might also be effective, although determination of this requires further experimentation for verification. Should rates as low as 0.5 parasitoid per plant per week combined with two buprofezin applications be effective, the cost would be approximately $0.19 per stem. If parasitoid releases could also be omitted for the 2-weeks when buprofezin was applied (reducing the numbers of weeks in which parasitoids would be released from 16 to 14), this cost would be

TABLE 6

Costs of Whitefly Control Treatments Applied in Greenhouses at Loosigian Farms, Methuen, Massachusetts, in 1997

Treatment/replicate	Total cost of parasitoids	Total cost of chemicals	Total cost	Number of single-stem poinsettias	Cost per single-stemmed plant
3 par., rep. 1	$1511.16	$0	$1511.16	1234	$1.22
3 par., rep. 2	$1235.39	$50[a]	$1285.39	1131	$1.14
2 par. + IGR, rep. 1	$910.44	$56.53[a]	$966.97	1250	$0.77
2 par. + IGR, rep. 2	$737.06	$6.53	$743.59	1020	$0.73
1 par. + IGR, rep. 1	$418.05	$6.53	$424.58	1126	$0.38
1 par. + IGR, rep. 2	$850.09	$46.21[a]	$896.3	2340	$0.38
Chemical control	$0	$115.42[a]	$115.42	800	$0.14

[a] Price includes at-harvest application of one or more Fulex Dithio smokes (Fuller Systems, Woburn, MA) as presale cleanup treatments for adult whiteflies.

further reduced to only $0.175 per stem, a price fully competitive with current pesticide-based control ($0.14, in this trial).

In conclusion, integration of buprofezin with low parasitoid release rates (one female parasitoid per plant per week) was as effective in suppressing whitefly populations as the high rate of *E. eremicus* (Table 3A), but had a much lower cost. Whitefly densities at harvest were below the informal standard of two whitefly nymphs + pupae per leaf that appears to be accepted by the poinsettia market in the northeastern United States.

Furthermore, in addition to being effective and affordable, we argue that integration of this IGR with parasitoid releases is less prone to control failures due to the development of pesticide resistance. Whereas resistance to insect growth regulators is possible (Horowitz and Ishaaya, 1994; Horowitz *et al.*, 1994; Ishaaya and Horowitz, 1995), it is likely only when IGRs are used as the exclusive means of whitefly control and when every generation of the whitefly is exposed to the chemical. Under the management program tested here, only one whitefly generation in three or four would be exposed to high levels of buprofezin, and every generation would be subject to mortality from parasitoid activities (parasitism and host feeding).

ACKNOWLEDGMENTS

We thank our cooperator, Greg Loosigian, for the use of his greenhouses. Funds from the Massachusetts IPM Program at the University of Massachusetts, Amherst and USDA/NRICGP Grant No. 9402481 supported this research.

REFERENCES

Boisclair, J., Brueren, G. J., and van Lenteren, J. C. 1990. Can *Bemisia tabaci* be controlled with *Encarsia formosa? SROP/WPRS Bull.* **13,** 32–35.

Byrne, D. N., Bellows, T. S., Jr., and Parrella, M. P. 1990. Whiteflies in agricultural systems. *In* "Whiteflies: Their Bionomics, Pest Status and Management" (D. Gerling, Ed.), pp. 227–261. Intercept, Ltd., Andover, UK.

Gerling, D., and Sinai, P. 1994. Buprofezin effects on two parasitoid species of whitefly (Homoptera: Aleyrodidae). *J. Econ. Entomol.* **87,** 842–846.

Helgesen, R. G., and Tauber, M. J. 1974. Biological control of greenhouse whitefly, *Trialeurodes vaporariorum* (Aleyrodidae: Homoptera), on short-term crops by manipulating biotic and abiotic factors. *Can. Entomol.* **106,** 1175–1188.

Hoddle, M. S., and Van Driesche, R. G. 1996. Evaluation of *Encarsia formosa* (Hymenoptera: Aphelinidae) to control *Bemisia argentifolii* (Homoptera: Aleyrodidae) on poinsettia (*Euphorbia pulcherrima*): A lifetable analysis. *Fla. Entomol.* **79,** 1–12.

Hoddle, M. S., and Van Driesche, R. G.. 1999. Evaluation of inundative releases of *Eretmocerus eremicus* and *Encarsia formosa* Beltsville strain in commercial greenhouses for control of *Bemisia argentifolii* on poinsettia stock plants. *J. Econ. Entomol.* **92,** 811–824.

Hoddle, M. S., Van Driesche, R. G., and Sanderson, J. P. 1997a. Biological control *of Bemisia argentifolii* (Homoptera: Aleyrodidae) on poinsettia with inundative releases of *Encarsia formosa* (Hymenoptera: Aphelinidae): Are higher release rates necessarily better? *Biol. Control* **10,** 166–179.

Hoddle, M. S., Van Driesche, R. G., and Sanderson, J. P. 1997b. Biological control *of Bemisia argentifolii* (Homoptera: Aleyrodidae) on poinsettia with inundative releases of *Encarsia formosa* "Beltsville strain" (Hymenoptera: Aphelinidae): Can parasitoid reproduction augment inundative releases? *J. Econ. Entomol.* **90,** 910–924.

Hoddle, M. S., Van Driesche, R. G., and Sanderson, J. P. 1998a. Biological control *of Bemisia argentifolii* (Hemiptera: Aleyrodidae) on poinsettia with inundative releases of *Eretmocerus eremicus* (Hymenoptera: Aphelinidae): Do release rates affect parasitism? *Bull. Entomol. Res.* **88,** 47–58.

Hoddle, M. S., Van Driesche, R. G., Elkinton, J. S., and Sanderson, J. P. 1998b. Discovery and utilization of *Bemisia argentifolii* patches by *Eretmocerus eremicus* and *Encarsia formosa* (Beltsville strain) in greenhouses. *Entomol. Exp. Appl.* **87,** 15–28.

Hoddle, M. S., Sanderson, J. P., and Van Driesche, R. G., 1999. Biological control of *Bemisia argentifolii* (Hemiptera: Aleyrodidae) on poinsettia with inunudative releases of *Eretmocerus eremicus* (Hymenoptera: Aphelinidae): Does varying the weekly release rate affect control? *Bull. Entomol. Res.* **89,** 41–51.

Hoddle, M. S., Van Driesche, R. G., Lyon, S., and Sanderson, J. P. 2000. Compatibility of insect growth regulators with *Eretmocerus eremicus* (Hymenoptera: Aphelinidae) for whitefly (Homoptera: Aleyrodidae) control on poinsettias: I. Laboratory Assays. *Biol. Control,* in press.

Horowitz, A. R., and Ishaaya, I. 1994. Managing resistance to insect growth regulators in the sweetpotato whitefly (Homoptera: Aleyrodidae). *J. Econ. Entomol.* **87,** 866–871.

Horowitz, A. R., Forer, G., and Ishaaya, I. 1994. Managing resistance in *Bemisia tabaci* in Israel with emphasis on cotton. *Pestic. Sci.* **42,** 113–122.

Ishaaya, I., and Horowitz, A. R. 1995. Pyriproxyfen, a novel insect growth regulator for controlling whiteflies: Mechanism and resistance management. *Pestic. Sci.* **43,** 227–232.

Jacobson, R. J., and Chambers, R. J. 1996. Control of glasshouse leafhopper (*Hauptidia maroccana:* Homoptera, Cicadellidae) within an IPM programme in protected tomatoes. *IOBC/WPRS Bull.* **19,** 67–70.

Jones, W. A., Ciomperlik, M. A., and Wolfenbarger, D. A.. 1998. Lethal and sublethal effects of insecticides on two parasitoids attacking *Bemisia argentifolii* (Homoptera: Aleyrodidae). *Biol. Control* **11,** 70–76.

Liu, T. X., Oetting, R. D., and Buntin, G. D. 1994. Evidence for interspecific competition between *Trialeurodes vaporariorum* (Westwood) and *Bemisia tabaci* (Gennadius) (Homoptera: Aleyrodidae) on some greenhouse grown plants. *J. Entomol. Sci.* **29,** 55–65.

Szabo, P., van Lenteren, J. C., and Huisman, P. W. T. 1993. Development time, survival and fecundity of *Encarsia formosa* on *Bemisia tabaci* and *Trialeurodes vaporariorum*. *SROP/WPRS Bull.* **16,** 173–176.

Van Driesche, R. G. 1983. The meaning of "percent parasitism" in studies of insect parasitoids. *Environ. Entomol.* **12,** 1611–1622.