

Efficacy Assessment of *Aphidius colemani* (Hymenoptera: Braconidae) for Suppression of *Aphis gossypii* (Homoptera: Aphididae) in Greenhouse-Grown Chrysanthemum

GISSELLA M. VÁSQUEZ, DAVID B. ORR,¹ AND JAMES R. BAKER

Department of Entomology, North Carolina State University, Gardner Hall, Box 7613, Raleigh, NC 27695

J. Econ. Entomol. 99(4): 1104–1111 (2006)

ABSTRACT To assess biological control as a management tool for the cotton aphid, *Aphis gossypii* Glover (Homoptera: Aphididae), the efficacy of *Aphidius colemani* Viereck (Hymenoptera: Braconidae) for suppression of *A. gossypii* in greenhouse-grown chrysanthemums, *Dendranthema grandiflora* (Tzvelev), was compared with a pesticide standard, imidacloprid (Marathon 1% G) and an untreated check. No significant differences were found between aphid populations in the two treatments. *A. colemani* and imidacloprid kept aphid numbers very low, with the correspondent aphid populations exhibiting very low intrinsic rates of increase ($r_m = -0.0369$ and $r_m = 0.0151$, respectively), in contrast to the exponential growth of aphid populations ($r_m = 0.1085$) observed on the untreated plants. Parasitism levels in *A. colemani* plots ranged from 48.93 to 83.38%. Esthetic damage parameters, including exuviae, honeydew, and sooty mold on leaves, were significantly different between treatments and untreated control, and damage levels were minimal with the insecticide treatment and natural enemy releases. The cost of *A. colemani* releases was 4.7 times greater than the cost of the imidacloprid treatment.

KEY WORDS augmentation biological control, inoculative releases, *Aphis gossypii*, *Aphidius colemani*, *Dendranthema grandiflora*

The cotton aphid, *Aphis gossypii* Glover (Hemiptera: Aphididae), occurs throughout the temperate, subtropic, and tropic zones of the world. It has a broad host range and has been discovered feeding on food, fiber, and ornamental crops in 88 plant families (Ebert and Cartwright 1997). This species is regarded as one of the most destructive aphids in the United States (Slosser et al. 1989), and it not only damages agricultural crops in the field but also is commonly found in greenhouses.

Chrysanthemum, *Dendranthema grandiflora* (Tzvelev), is one of the major floricultural crops grown in greenhouses throughout the world, and the United States is among the top five major producers (Parrella et al. 1999). In 2004 in the United States, both cut and potted chrysanthemums were sold with a value of approximately \$228.7 million (Agricultural Statistics Board, NASS, USDA 2005). Aphids are important pests of chrysanthemum, causing direct damage through feeding, which results in wilting, leaf distortion, and transmission of several viruses; and indirect damage through physical contamination with aphid exuviae and honeydew that is also a nutrient source for sooty mold (Agrios 1988). *A. gossypii* is among the most serious pests of greenhouse-grown chrysanthemum (Guldemon et al. 1994, Storer and van Emden 1995,

Bethke et al. 1998). Because of the parthenogenetic nature of the aphid's reproductive cycle and the relatively stable and favorable growing environment for both insect and host plant under greenhouse production conditions, high cotton aphid densities on chrysanthemum are both an esthetic and a crop production problem during any season at any stage in the cropping cycle (Bethke et al. 1998), being particularly difficult to control at the reproductive stage when noticeable, dense aphid clumps in flowers greatly reduce the value of the crop. In addition, the importance of cotton aphids to chrysanthemum production has increased because they have developed resistance to various classes of insecticides, including organophosphates and carbamates (Scopes and Ledieu 1980, Furk and Vedjhi 1990, Furk and Hines 1993, Herron et al. 2001).

Biological control, as a component within integrated pest management (IPM) programs, is a powerful pest control option in situations where chemical control is insufficient, impossible, or undesired (van Lenteren 2000). The majority of successful cases of biological control of insect pests in greenhouses are reported in vegetable crops, and few studies have examined the use of biological control in ornamental crops (Heinz and Parrella 1990). *Aphidius colemani* Viereck (Hymenoptera: Braconidae) is an aphid parasitoid that has been reared on dozens of aphid species and is considered a potentially effective biological

¹ Corresponding author, e-mail: david_orr@ncsu.edu.

control agent against several economically important pests (van Schelt 1994, Messing and Rabasse 1995), including *A. gossypii* (van Steenis 1995, van Steenis and El-Khawass 1995, Heinz 1998, Jacobson and Croft 1998). The effectiveness of inoculative releases of *A. colemani* to achieve suppression of cotton aphid attacking greenhouse-grown chrysanthemums was examined in this study as a first step to control one of the major insect pests of this crop. Marathon 1% G (imidacloprid) was evaluated as an insecticide standard. This chloronicotinyl insecticide is generally recommended for aphid control because it can effectively reduce aphid survival and has long-lasting residual activity (Pasian et al. 1997, Nauen et al. 1998). Most studies on aphid control in greenhouse ornamental crops have examined the efficacy of chemical or biological control separately, and few studies have compared both types of control directly. Therefore, the goal of this study was to compare cotton aphid suppression on greenhouse-grown chrysanthemum achieved by *A. colemani* releases and imidacloprid.

Materials and Methods

Insect Cultures. The aphids used in this study were collected on 13 March 2001 from gardenias, *Gardenia* sp., from a commercial greenhouse (Oakmont Nursery, Chatham County, North Carolina) and on 5 April 2001 from 'Kory' chrysanthemums from a local grocery store (Harris Teeter, Wake County, North Carolina). Species identification was confirmed by David Stephan at North Carolina State University, Raleigh, NC. Laboratory cultures of aphids were maintained on Kory chrysanthemums in an environmental chamber set at $23 \pm 2^\circ\text{C}$, 40–70% RH, and a photoperiod of 16:8 (L:D) h. *A. colemani* produced by Syngenta Bioline (Little Clacton, Essex, England) and sold as Aphiline c 500, was purchased from The Green Spot Ltd. (Nottingham, NH). From all the parasitoid species with potential to control cotton aphid, *A. colemani* was selected based on reports of its ability to provide successful biological control of aphids infesting greenhouse crops (van Schelt 1994).

Plant Cultures. Kory chrysanthemum is among the cultivars commonly grown by producers of spray pot mums and is susceptible to cotton aphid. Plants were grown in a greenhouse in the North Carolina State University Phytotron under controlled conditions (22/18°C day/night, 40–83% RH, and a photoperiod of 16:8 [L:D] h) to maintain them in a vegetative state and to keep them pest free. Three hundred rooted cuttings (Yoder Brothers Inc., Barberton, OH) were transplanted in groups of three into pots (15.3 cm in diameter) filled with Mix No. 3-S (Conrad Fafard, Inc., Agawam, MA) and fertilized daily with a 10.2-1-10.7 (N-P-K) solution (106.23 mg N/liter). Fourteen days after transplanting, the terminal 2 to 3 cm of each stem was removed or "pinched" off to increase branching, as in commercial production. Plants were held in the phytotron for 6 wk until needed for the experiment. At the beginning of the study, the plants had a total of

63.5 ± 0.7 leaves (mean \pm SE), a height of 21.4 ± 0.1 cm (mean \pm SE), and they had not initiated flowering.

Experimental Design. A portion of a greenhouse on the North Carolina State University campus was subdivided into three bays of 2.15 by 6.10 m with Econet-S (Ludvig Svensson, Inc., Charlotte, NC) as screening material (0.14- by 0.14-mm hole size) allowing air circulation through the bays but preventing movement of aphids between bays. The experiment was set up as a randomized complete block with selective placement of one treatment, and repeated three times, with each trial taking 35 d. Blocking was done across bays to account for the temperature gradient from the air inlet to the air outlet. Within each block the parasitoid treatment was assigned to the same bay. The insecticide treatment and control were randomly assigned to the other two bays within each block. The parasitoid releases were always made in a single bay because of the need to enclose the area. However, in each of the three trials, the bay used for release of parasitoids was chosen randomly. Within a trial, there were 10 pots (three plants per pot) for each treatment in each block. Pots were spaced 30 cm apart to provide 0.45 m² of bench space per pot. Trials were performed from 2 June to 6 July 2001, 14 July to 17 August 2001, and 25 August to 28 September 2001.

Five apterous adult aphids per pot were placed on the top leaves 48 h before treatment application. The two treatments in this study were 1) three weekly releases of *A. colemani*, by using producer recommended rates and methods (i.e., five adults per square meter at sundown), and 2) one application of imidacloprid (Marathon 1% G, Olympic Horticultural Products, Mainland, PA) used as a standard insecticide treatment (14 mg active ingredient per pot). Untreated plants acted as controls. Two yellow sticky traps (20 by 15 cm) were placed between control and pesticide treatments to reduce the likelihood of aphid migration. Plants were irrigated twice daily and fertilized with 7.4 ml Osmocote 14-14-14 (Scotts Sierra Horticultural Products Company, Marysville, OH) per pot. The greenhouse was sprayed with fluralinate (Mavrick Aqua Flow, Sandoz Agro, Inc., Des Plaines, IL) at a rate of 2.44 ml/liter between trials to eliminate undesired insects.

Data Collection and Statistical Analyses. Aphids were counted with the aid of a magnifier glass (5 \times magnification) every 7 d for a period of 35 d after the initial release. All aphids on each of three randomly selected leaves per pot were counted. Sample leaves were from top, middle, and bottom sections of the plant to provide an average measure of aphid density per plant. For each sampling date, 90 leaves per treatment (30 for each plant section) were sampled. The numbers of mummified aphids per leaf also were counted to estimate parasitism levels. Plant height and number of leaves were recorded on day 1 and day 35. To evaluate plant damage, plants were visually inspected within each section, and the proportion of plant foliage that displayed the presence of exuviae adhered to honeydew, and sooty mold growth was estimated on days 21, 28, and 35. Environmental con-

ditions were monitored using Hobo H8 temperature and humidity data loggers (Onset Computer Corp., Pocasset, MA). All means reported are followed by standard errors.

For each sample date, the number of aphids per leaf within each plant section and the number of aphids per leaf over all plant sections were transformed to natural logarithms [$\log_e(x + 1)$]. Data were analyzed using the General Linear model Procedure (PROC GLM). Trials and blocks within trial were considered random effects; treatments, days, and plant sections were considered fixed. In addition, the repeated measures factor day was considered a subplot factor (and tested against trial by day), and plant section was treated as a sub-subplot factor. TEST statements were included to generate appropriate F-tests. Depending on the presence of interactions, the appropriate sets of treatment and control means were separated using the least significant difference (LSD) test at a significance level of 0.05 (SAS Institute 1999).

When $\log_e(x)$ increases linearly with time, the slope of the regression line estimates the population growth rate (r_m) in the equation $N_t = N_0 e^{r_m t}$ (Vehrs et al. 1992). To analyze *A. gossypii* population growth, the mean number of aphids per leaf over all plant sections for each sample date was transformed to natural logarithms [$\log_e(x + 0.15)$], and the mean and standard error of the transformed data were plotted against time over all sample dates. Rates of r_m were estimated by averaging the slopes of growth curves obtained from each of the treatment or control blocks (nine blocks per treatment or control). Slopes were compared between untreated *A. gossypii* populations and *A. gossypii* populations controlled by *A. colemani* and imidacloprid with analysis of variance (ANOVA) by using the GLM procedure, with trials considered random. Means were separated using the LSD test at a significance level of 0.05 (SAS Institute 1999).

Levels of parasitism were analyzed for each sample date, for each plant section, and over all sections with ANOVA by using the GLM procedure. Percentages of parasitized aphids were arcsine transformed (arcsine x^{-2}) for analysis. The relationship between the percentage of parasitized aphids per leaf and the mean number of aphids per leaf on each plant section was analyzed.

Plant height and number of leaves per plant were compared between treatments at the beginning and at the end of each trial with ANOVA. The analysis of aphid damage used an arcsine transformation (arcsine x^{-2}) for the percentage of the plants in each pot covered by honeydew, cast skins, and sooty mold. The data were subjected to ANOVA by using the GLM procedure and mean separation by least-squares means to determine differences between aphid damage at a significance level of 0.05 (SAS Institute 1999).

Mean daily temperature and mean relative humidity recorded for each bay per trial were compared using the Mixed Procedure (SAS Institute 1999). Mean temperature and mean relative humidity data were plotted for each hour of the day. These means were obtained by averaging temperature and relative

humidity for each specific hour for the duration of the experiment. Maximum and minimum daily temperature and maximum and minimum daily relative humidity also were recorded for each bay per trial.

For both treatments, product prices were recorded and application costs were estimated based on necessary equipment and training required (see Table 2 for detailed information on costs). These data were used to compare natural enemy releases and insecticide application costs. Costs were extrapolated for a 305-m² commercial greenhouse operation.

Results

Aphid populations differed significantly between treatments (*A. colemani* and imidacloprid) and the untreated control when averaging mean number of aphids per leaf across plant sections ($F = 110.88$; $df = 2, 4$; $P = 0.0003$). We also found a significant treatment by sample date interaction ($F = 36.27$; $df = 8, 16$; $P < 0.0001$), with higher *A. gossypii* numbers observed in the untreated control than in both treatments on every sample date (Fig. 1), except for day 7 for which *A. colemani* treatment (1.11 ± 0.12 aphids per leaf), imidacloprid treatment (0.04 ± 0.02 aphids per leaf), and untreated plants (3.84 ± 0.34 aphids per leaf) all differed (LSD means separation test; $P < 0.05$). *A. gossypii* numbers were kept very low in both natural enemy and insecticide treatments from the beginning through the last day of the study. Five weeks after treatment applications the mean number of aphids per leaf was 0.34 ± 0.06 and 0.22 ± 0.03 aphids for *A. colemani* and imidacloprid treatments, respectively, compared with 76.63 ± 5.6 aphids per leaf on untreated plants.

Mean number of aphids per leaf averaged across all sample dates was different among bottom, middle, and top plant sections ($F = 684.02$; $df = 2, 60$; $P < 0.0001$). Differences in mean number of aphids per leaf (averaged across all sample dates) between treatments and control were consistent on the bottom ($F = 175.49$; $df = 2, 4$; $P = 0.0001$), middle ($F = 312.11$; $df = 2, 4$; $P < 0.0001$), and top ($F = 17.11$; $df = 2, 4$; $P = 0.0110$) plant sections. By the end of the study, the mean number of aphids per leaf throughout the bottom, middle, and top sections of untreated plants was 187.29 ± 8.31 , 31.63 ± 1.86 , and 0.98 ± 0.68 aphids, respectively. *A. colemani* and imidacloprid treatments reduced the numbers of *A. gossypii* in the bottom plant section, so that at the end of the experiment only 0.68 ± 1.18 and 0.44 ± 0.07 aphids per leaf were recorded, respectively. Likewise, *A. colemani* and imidacloprid treatments reduced the numbers of *A. gossypii* throughout the middle plant section, so that by the end of the study, the mean numbers of aphids per leaf were 0.27 ± 0.05 and 0.17 ± 0.04 , respectively. Aphid densities were never high on the top plant section, and mean numbers of aphids per leaf by the end of the study were 0.08 ± 0.03 and 0.04 ± 0.02 for *A. colemani* and imidacloprid treatments, respectively.

The slopes of population growth curves (estimates of r_m) differed between treatments and untreated

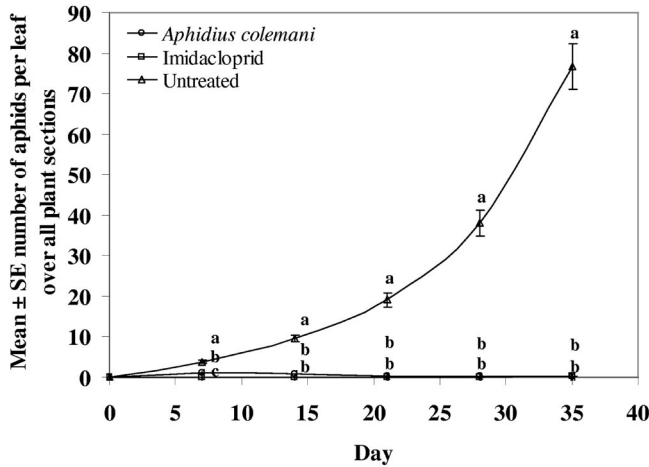


Fig. 1. Densities (mean ± SE) of *A. gossypii* on greenhouse-grown chrysanthemum treated with either *A. colemani* releases (○) or imidacloprid application (□) and compared with untreated control (△). Mean separation by LSD. Values on the same date followed by the same letter are not significantly different ($P < 0.05$, $\alpha = 0.05$, $n = 90$). Mean separation based on natural logarithm-transformed data.

plants ($F = 41.04$; $df = 2, 4$; $P = 0.0022$) and were the largest for untreated plants ($r_m = 0.1085 \pm 0.0073$), smaller for imidacloprid-treated plants ($r_m = 0.0151 \pm 0.0047$), and the smallest for *A. colemani* treated plants ($r_m = -0.0369 \pm 0.0084$) (Fig. 2).

Aphids with signs of parasitism (tan colored with bloated appearance) were observed on the first evaluation day. Mummified aphids were evident within 14 d of parasitoid release. Levels of parasitism averaged across all plant sections differed significantly across days of the experiment ($F = 6.12$; $df = 4, 8$; $P = 0.0148$), increasing considerably during the first 3 wk of the experiment. The initial parasitism level recorded was $48.93 \pm 4.33\%$ and increased to $66.46 \pm 3.92\%$ on day 14. Three weeks after the first parasitoid release, parasitism level was $83.38 \pm 2.15\%$ and re-

mained fairly constant until the end of the experiment, when parasitism level was $83.00 \pm 3.11\%$. No significant differences in parasitism levels were found among plant sections ($F = 0.46$; $df = 2, 4$; $P = 0.6603$). At the end of the experiment, the mean number of aphids and mummies combined per leaf was 3.81 ± 3.67 , 1.70 ± 1.43 , and 0.69 ± 0.96 for bottom, middle, and top plant sections, respectively. Based on the parasitism levels recorded, the mean number of non-mummified aphids per leaf estimated for each plant section was always less than 1, with 0.68 ± 1.67 , 0.27 ± 0.51 and 0.08 ± 0.27 aphids per leaf for bottom, middle, and top plant sections, respectively. No significant relationship between mean number of aphids per leaf and parasitism level was found (Pearson correlation coefficient = 0.0104 ; $P = 0.9084$).

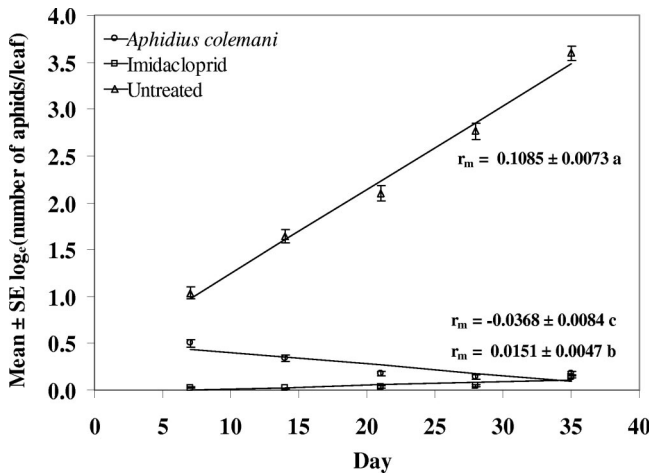


Fig. 2. Population growth rates (mean ± SE) of *A. gossypii* on greenhouse-grown chrysanthemum treated with either *A. colemani* releases (○) or imidacloprid application (□) and compared with untreated control (△). Mean separation by LSD. Slopes with the same letter are not significantly different ($P < 0.05$, $\alpha = 0.05$, $n = 9$).

Table 1. *A. gossypii* damage assessment on chrysanthemums treated with a parasitic wasp, *A. colemani*, and a systemic chloronicotinyl insecticide, imidacloprid

Treatment	Mean \pm SE exuviae and honeydew (%)			Mean \pm SE sooty mold (%)		
	day 21	day 28	day 35	day 21	day 28	day 35
<i>A. colemani</i>	2.07 \pm 0.23b	2.64 \pm 0.26b	3.50 \pm 0.31b	0.04 \pm 0.03b	0.04 \pm 0.03b	0.06 \pm 0.03b
Imidacloprid	0.00c	0.01 \pm 0.01c	0.11 \pm 0.04c	0.00b	0.00b	0.00b
Untreated	14.93 \pm 0.86a	30.9 \pm 1.61a	63.09 \pm 1.59a	1.81 \pm 0.33a	5.35 \pm 0.60a	11.96 \pm 0.79a

Mean separation by least-squares means. Means within a column followed by different letters are significantly different at $\alpha = 0.05$, $P < 0.05$, $n = 90$.

Indirect cosmetic damage was evaluated as percentage of the plant covered with exuviae adhered to honeydew, and percentage of the plant with black sooty mold growth.

The plants grew from 21.4 ± 0.1 cm with 63.5 ± 0.7 leaves on day 0 to 39.2 ± 0.2 cm with 125.0 ± 1.7 leaves on day 35. No significant differences in plant height were found between treatment and control plants either on day 0 ($F = 0.46$; $df = 2, 4$; $P = 0.6600$) or on day 35 ($F = 0.44$; $df = 2, 4$; $P = 0.6737$). No significant differences in total number of leaves per plant were found between treatment and control plants on day 0 ($F = 1.09$; $df = 2, 4$; $P = 0.4194$) or on day 35 ($F = 1.78$; $df = 2, 4$; $P = 0.2803$). Plant height and number of leaves per plant consistently increased, indicating no plant growth effects from any of the treatments.

Levels of esthetic damage differed between treatments and untreated control. Aphid damage to chrysanthemums is caused indirectly by damage from aphid exuviae and honeydew secretion. The presence of exuviae adhered to honeydew on plants differed significantly between treatments and untreated plants ($F = 1680$; $df = 2, 4$; $P < 0.0001$). The greatest percentage of exuviae and honeydew covering the plants for each evaluation date was always observed on untreated plants (Table 1). Sooty mold growth differed significantly between treatments and untreated plants ($F = 779.86$; $df = 2, 4$; $P < 0.0001$), with levels of sooty mold covering the plants always the greatest for untreated plants (Table 1). No significant differences were found for presence of sooty mold on both *A. colemani* and imidacloprid-treated plants (least-squares mean separation test; $P < 0.05$).

The mean daily temperature and the mean daily relative humidity recorded over all trials was $26.1 \pm 0.25^\circ\text{C}$ and $75.8 \pm 0.91\%$ RH, respectively. Although significant differences were found between bays for mean daily temperature ($F = 35.89$; $df = 2, 204$; $P < 0.0001$) and mean relative humidity ($F = 15.57$; $df = 2, 186$; $P < 0.0001$), these differences were not $>0.9^\circ\text{C}$ and 3.1% RH for temperature and relative humidity, respectively. Vásquez (2001) provides detailed information on differences in temperature and relative humidity among bays. Environmental conditions were considerably similar between bays, and differences in temperature (0.4 – 0.9°C) and relative humidity (1.5 – 3.1% RH) seemed to have no major biological impact on insect populations and plant material.

Costs associated with *A. gossypii* biological control were compared with costs associated with imidacloprid application, as extrapolated for 305 m^2 of greenhouse space (Table 2). Three *A. colemani* releases would be necessary for suppression of *A. gossypii* dur-

ing the crop cycle at an estimated price of \$112.78 per release. Product total cost, multiplied by the total number of releases, and with labor cost (U.S. Department of Labor 2004) added, yielded a total cost of \$342.65 for a biological control program with *A. colemani*. Aphid control with imidacloprid involved only one application during the crop cycle at a cost of \$27.22 per application. The total cost of this insecticide-based control, including labor cost, personal protective equipment cost, safety training cost, and private pesticide applicator license cost, was \$72.44. To suppress cotton aphid on chrysanthemums maintained in a vegetative growth stage for 5 wk, chemical control costs about \$0.16 per pot, and biological control costs \$0.76.

Discussion

In this study, we focused on aphid management in the vegetative period of plant growth between pinch

Table 2. Product and application costs of *A. colemani* and imidacloprid for control of cotton aphid on chrysanthemums grown in a 305-m^2 greenhouse

	Imidacloprid	<i>A. colemani</i>
Product cost	\$19.60/453.6 g	\$30.95/bottle of 500 mummies
Product application rate	1.4 g/pot	5 adults/ m^2
Product total cost (305-m^2 greenhouse)	\$27.22 ^a	\$338.34 ^b
Labor cost ^c	\$6.72	\$4.32
Other costs ^d	\$38.50	N/A ^e
Total cost	\$72.44	\$342.65

^a Cost of applying imidacloprid in a 305-m^2 greenhouse with 15 benches (15 m^2 each) with 30 pots per bench (0.5 m^2 of bench space per pot), for a total of 450 pots.

^b Cost of releasing *A. colemani* with a release rate of 2.82 bottles per week for 3 wk in a greenhouse with 225 m^2 of bench space. Shipping and handling cost (\$25.50) was added to product cost per weekly shipment. Number of bottles needed per week was estimated using a 64.1% *A. colemani* emergence rate and 623 mummies as the total number of mummies received per bottle.

^c Labor cost based on a \$8.23/h wage reported for crop, nursery, and greenhouse farmworkers and laborers (U.S. Department of Labor, Bureau of Labor Statistics, Occupational Employment Statistics 2004). Estimated time for imidacloprid application in 450 pots is 49 min. Estimated time for *A. colemani* releases in an area of 225 m^2 (greenhouse bench space) is 10.5 min per release.

^d Costs include personal protective equipment cost, safety training cost, and private pesticide applicator license fee.

^e Not applicable.

and disbud. It is critical for growers to completely control aphids at this stage to prevent them from reaching the flowers, where they are difficult to control and reduce the crop value by affecting its visual appeal. Pest populations must be kept at very low levels because flower growers cannot tolerate damage; thus, the concept of using natural enemies for pest control in ornamentals has met with resistance (Scopes and Ledieu 1980). However, the need for early control to avoid aphid damage has led to a repeated heavy use of insecticides. This is not only inconsistent with IPM but also contributes to development of aphid pesticide resistance, which renders chemicals less reliable.

Both natural enemy releases and insecticide application suppressed *A. gossypii*. The systemic insecticide imidacloprid (Marathon 1% G) generally provides good-to-excellent long-term control of whiteflies, aphids, and other piercing-sucking pests, and although it can be very effective and new application methods may further increase the effectiveness of low rates (Pasian et al. 1997), its application to potted plants is in some instances labor-intensive, and pesticide exposure hazards and worker reentry restrictions are still present. *A. colemani* has the potential to reduce aphid populations compared with this standard insecticide treatment. It seems that this parasitoid species is able to find and attack low densities of aphids and probably exhibits a type 2 or logistic functional response (van Steenis and El-Khawass 1995). Therefore, the best strategy would be to release it as soon as possible (Rochat 1997) before aphid densities become too high. At higher aphid densities a strong numerical response is present through aggregation at places with high aphid densities. The combination of both responses may account for the successes obtained with biological control of aphids with *A. colemani* (van Steenis and El-Khawass 1995). Similarly in our experiment, augmentative releases of *A. colemani* provide control of low aphid densities. *A. colemani* is capable of spreading across 12,768 m² of greenhouse space, and at a recommended spacing of release points, complete coverage of a chrysanthemum range is feasible (Heinz 1998).

Environmental conditions were similar between bays, and these slight differences in temperature (0.4–0.9°C) and relative humidity (1.5–3.1% RH) had little biological impact on insect populations and plant material. This assumption allowed us to obtain comparable results on insect species performance between bays and trials. When temperatures exceed 25°C, *A. colemani* develops atypically, and when temperatures increase to 30°C, failure of parasitism is frequent (Guenaooui 1991). Maximum daily temperatures were always above 35°C, and minimum daily temperatures were ≈19°C. Despite of the negative physiological effects that high temperatures may have on *A. colemani*, it seems that the parasitoid performance was not greatly affected, because aphid levels were always low.

A. gossypii became more abundant on the lower section of the plants, particularly the bottom plant

section. Similar observations were reported by Vehrs et al. (1992) and Storer and van Emden (1995) in different cultivars, whereas Guldemond et al. (1998) found that *A. gossypii* was also abundant on the top part of young plants. Differences in aphid distribution within the plant can be related to levels of fertilization, with aphids distributing predominantly in areas with higher nitrogen content (Chau et al. 2005).

The intrinsic rate of increase reported in our study for *A. gossypii* ($r_m = 0.11 \pm 0.0073$) is low compared with the rates reported by the same species reared on other chrysanthemum cultivars by Vehrs et al. (1992) ($r_m = 0.17 \pm 0.0080$), Storer and van Emden (1995) (from $r_m = 0.14 \pm 0.0144$ to $r_m = 0.27 \pm 0.0109$), Bethke et al. (1998) (from $r_m = 0.16 \pm 0.0101$ to $r_m = 0.22 \pm 0.0180$), and Guldemond et al. (1998) ($r_m = 0.35 \pm 0.0080$). These differences can be attributed to host plant factors affecting growth rate, i.e., plant suitability as a host, plant growth stage, and physiology, and its effects on aphid distribution patterns. This is consistent with the fact that different cultivars show differential resistance to aphids, affecting aphid performance and limiting damaging populations. High levels of nitrogen also may decrease aphid performance (Bethke et al. 1998), but in our study the levels of N in the fertilizer were no >150 mg N/liter. The period used in our study to estimate aphid growth rate was longer than in previous studies, and although populations of *A. gossypii* seemed to grow exponentially on untreated plants throughout the experiment, this also may be another factor affecting our estimation of growth rate. Temperature also can affect the rate of increase of *A. gossypii*, with the highest rate of increase of *A. gossypii* populations on cotton observed at 25°C (Xia et al. 1999). In our study, aphid growth was controlled with both natural enemy and insecticide treatments, and growth rates were minimal, indicating that the growth potential of the pest was clearly affected and that their impacts were not only effective but also consistent through time. Among commercial chrysanthemum cultivars, Kory is probably less susceptible to aphids than other cultivars, but how these differences in susceptibility may affect biological and chemical control is unclear. Resistant cultivars may complement the activity of the parasitoid in reducing the population density of sucking pests, and natural enemy activity on partially resistant cultivars can be greater than would be expected (van Emden 1986).

The total costs associated with *A. colemani* biological control were 4.7 times greater than the costs associated with the insecticide treatment. However, *A. colemani* is safe both to agricultural workers and to the environment, and it has a small chance of causing resistance in aphids. Van Lenteren (2000) provides specific advantages of biological pest control in greenhouses, including the lack of phytotoxic effects on young plants and premature abortion of flowers. Natural enemy release takes less time and is more pleasant than applying chemicals in humid and warm greenhouses, and selection of chemicals is limited for some key pests because of pesticide resistance. Improved

mass-rearing techniques, improved mass-production with special attention to product quality control, and availability of efficacy data on augmentative releases could make commercially available natural enemies not only more reliable but also more reasonably priced. Moreover, aphid control could combine the use of natural enemies with compatible pesticides such as insect growth regulators (Hoddle et al. 2001, Sechser et al. 2002) to obtain a higher reduction in levels of aphid infestation as has been shown for the cabbage aphid, *Brevicoryne brassicae* (L.) (Acheampong and Stark 2004). Also, early *A. colemani* releases to prevent aphid outbreaks can be compatible with recommended biological control practices for the Western flower thrips, *Frankliniella occidentalis* (Per-gande), one of the most damaging and difficult pest to control in greenhouse flower crops.

Levels of esthetic damage were minimal with the insecticide treatment and with natural enemy releases, but to obtain comparable control, it is important to release natural enemies early within the crop cycle and to use an adequate natural enemy release rate. Considerable variation exists among introduction methods and release rates for the various commercially available parasitoids (Parrella and Heinz 1998). In this study, low densities of cotton aphid (25.2 adults/m² 48 h before release) were suppressed with a parasitoid release rate of 5 adults/m² and following producer recommendations.

Production of chrysanthemums that will meet grower esthetic standards can be achieved by inoculative releases of *A. colemani*, providing an effective *A. gossypii* management alternative to traditional insecticide control. This option could be even more appealing if costs were reduced. In addition, pests continue to develop resistance to chemical pesticides, particularly in greenhouses, where repeated pesticide applications exert strong selective pressure on whole insect populations (van Lenteren 2000). Pest resistance is unlikely to occur with the use of parasitoids. Growers of floricultural crops can consider the use of *A. colemani* releases as an effective, safe, and reliable way for suppression of *A. gossypii* in greenhouses.

Acknowledgments

We thank Ludvig Svensson, Inc. for providing the Econet-S screening material and the North Carolina State University Phytotron for space and cultural requirements for growing the experimental chrysanthemum plants. Appreciation is also extended to Cavell Brownie for statistical assistance. We thank Christine Casey and Steve Bambara for helpful comments on the manuscript. We are grateful to Ellen Reeves, Melinda Gibbs, and Lesley Canup for technical assistance.

References Cited

Acheampong, S., and J. S. Stark. 2004. Can reduced rates of pymetrozine and natural enemies control the cabbage aphid, *Brevicoryne brassicae* (Homoptera: Aphididae), on broccoli? *Int. J. Pest Manage.* 50: 275-279.

- Agricultural Statistics Board, NASS, U.S. Dep. Agric. 2005. Floriculture Crops 2004 Summary. April 2005. (<http://usda.mannlib.cornell.edu/reports/nassr/other/zfc-bb/floran05.pdf>).
- Agrios, G. N. 1988. *Plant pathology*, 3rd ed. Academic, San Diego, CA.
- Bethke, J. A., R. A. Redak, and U. K. Schuch. 1998. Melon aphid performance on chrysanthemum as mediated by cultivar, and differential levels of fertilization and irrigation. *Entomol. Exp. Appl.* 88: 41-47.
- Chau, A., K. M. Heinz, and F. T. Davies, Jr. 2005. Influences of fertilization on *Aphis gossypii* and insecticides usage. *J. Appl. Entomol.* 129: 176-184.
- Ebert, T. A., and B. Cartwright. 1997. Biology and ecology of *Aphis gossypii* Glover (Homoptera: Aphididae). *Southwest. Entomol.* 22: 116-153.
- Furk, C., and S. Vedjhi. 1990. Organophosphorus resistance in *Aphis gossypii* (Hemiptera: Aphididae) on chrysanthemum in the UK. *Ann. Appl. Biol.* 116: 557-561.
- Furk, C., and C. M. Hines. 1993. Aspects of pirimicarb resistance in the cotton and melon aphid, *Aphis gossypii* Glover (Homoptera: Aphididae). *Ann. Appl. Biol.* 123: 9-17.
- Guenaoui, Y. 1991. Role of temperature on the host suitability of *Aphis gossypii* Glover (Hom.: Aphididae) for the parasitoid *Aphidius colemani* Viereck (Hym.: Aphididae). *In Insect Parasitoids. 4th European Workshop*, 3-5 April 1991, Perugia. *REDIA* 74: 163-165.
- Guldmond, J. A., W. T. Tigges, and P.W.F. De Vrijer. 1994. Host races of *A. gossypii* (Homoptera: Aphididae) on cucumber and chrysanthemum. *Environ. Entomol.* 23: 1235-1240.
- Guldmond, J. A., W. J. van den Brink, and E. den Belder. 1998. Methods of assessing population increase in aphids and the effect of growth stage of the host plant on population growth rates. *Entomol. Exp. Appl.* 86: 163-173.
- Heinz, K. M. 1998. Dispersal and dispersion of aphids (Homoptera: Aphididae) and selected natural enemies in spatially subdivided greenhouse environments. *Environ. Entomol.* 27: 1029-1038.
- Heinz, K. M., and M. P. Parrella. 1990. Biological control of insect pests on greenhouse marigolds. *Environ. Entomol.* 19: 825-835.
- Herron, G. A., K. Powisand, and J. Rophail. 2001. Insecticide resistance in *Aphis gossypii* Glover (Hemiptera: Aphididae), a serious threat to Australia cotton. *Aust. J. Entomol.* 40: 85-91.
- Hoddle, M. S., R. G. Van Driesche, S. M. Lyon, and J. P. Sanderson. 2001. Compatibility of insect growth regulators with *Eretmocerus eremicus* (Hymenoptera: Aphelinidae) for whitefly (Homoptera: Aleyrodidae) control on poinsettias. *Biol. Control* 20: 122-131.
- Jacobson, R. J., and P. Croft. 1998. Strategies for the control of *Aphis gossypii* Glover (Hom.: Aphididae) with *Aphidius colemani* Viereck (Hym.: Braconidae) in protected cucumbers. *Biocontrol Sci. Technol.* 8: 377-387.
- Messing, R. H., and J. M. Rabasse. 1995. Oviposition behavior of the polyphagous aphid parasitoid *Aphidius colemani* Viereck (Hymenoptera: Aphididae). *Agric. Ecosyst. Environ.* 52: 13-17.
- Nauen, R., K. Tietjen, K. Wagner, and A. Elbert. 1998. Efficacy of plant metabolites of imidacloprid against *Myzus persicae* and *Aphis gossypii* (Homoptera: Aphididae). *Pestic. Sci.* 52: 53-57.
- Parrella, M. P., and K. M. Heinz. 1998. Parasitoids for control of greenhouse pests, pp. 27-89. *In R. L. Ridgway, M. P. Hoffmann, M. N. Inscoc, and C. S. Glenister [eds.], Mass-reared natural enemies: application, regulation, and*

- needs. Thomas Say Publications in Entomology: Proceedings. Entomological Society of America, Lanham, MD.
- Parrella, M. P., L. S. Hansen, and J. C. van Lenteren. 1999. Glasshouse environments, pp. 819–839. *In* T. S. Bellows and T. W. Fisher [eds.], Handbook of biological control. Academic, New York.
- Pasian, C. C., R. K. Lindquist, and D. K. Struve. 1997. A new method of applying imidacloprid to potted plants for controlling aphids and whiteflies. *HortTechnology* 7: 265–269.
- Rochat, J. 1997. Delayed effects in aphid parasitoid systems: consequences for evaluating biological control species and their use in augmentation strategies. *Entomophaga* 42: 201–213.
- SAS Institute. 1999. SAS online document, version 8.1. SAS Institute, Cary, NC.
- Scopes, N.E.A., and M. S. Ledieu. 1980. Integrated pest control on chrysanthemums and other flower crops. *Sci. Hortic.* 31: 48–53.
- Sechser, B., B. Reber, and F. Bourgeois. 2002. Pymetrozine: selectivity spectrum to beneficial arthropods and fitness for integrated pest management. *J. Pest Sc.* 75: 72–77.
- Slosser, J. E., W. E. Pinchak, and D. R. Rummel. 1989. A review of known and potential factors affecting the population dynamics of the cotton aphid. *Southwest. Entomol.* 14: 302–313.
- Storer, J. R., and H. F. van Emden. 1995. Antibiosis and antixenosis of chrysanthemum cultivars to the aphid *Aphis gossypii*. *Entomol. Exp. Appl.* 77: 307–314.
- U.S. Department of Labor, Bureau of Labor Statistics, Occupational Employment Statistics. 2004. Occupational employment and wages, November 2004. (http://www.bls.gov/oes/2004/november/oes_45Fa.htm).
- van Emden, H. F. 1986. The interaction of plant resistance and natural enemies: effects on populations of sucking pests, pp. 138–150. *In* D. J. Boethel and R. D. Eikenbary [eds.], Interactions of plant resistance and parasitoids and predators of insects. Ellis Horwood Series in Entomology. Chichester, United Kingdom.
- van Lenteren, J. C. 2000. A greenhouse without pesticides: fact or fantasy. *Crop Prot.* 19: 375–384.
- van Schelt, J. 1994. The selection and utilization of parasitoids for aphid control in glasshouses. *Proceedings of the Section Experimental and Applied Entomology of The Netherlands Entomological Society (N.E.V.)* 5: 151–157.
- van Steenis, M. J. 1995. Evaluation of four aphidiine parasitoids for biological control of *Aphis gossypii*. *Entomol. Exp. Appl.* 75: 151–157.
- van Steenis, M. J., and K.A.M. H. El-Khawass. 1995. Behaviour of *Aphidius colemani* searching for *Aphis gossypii*: functional response and reaction to previously searched aphid colonies. *Biocontrol Sci. Technol.* 5: 339–347.
- van Steenis, M. J., and K.A.M.H. El-Khawass. 1996. Different parasitoid introduction schemes determine the success of biological control of *Aphis gossypii* with the parasitoid *Aphidius colemani*. *Bull. IOBC/WPRS* 19: 159–162.
- Vásquez, G. M. 2001. Assessment of quality and efficacy of selected commercially available biological control agents for greenhouse pest management. M.S. thesis, North Carolina State University, Raleigh, NC.
- Vehrs, S.L.C., G. P. Walker, and M. P. Parrella. 1992. Comparison of population growth rate and within-plant distribution between *Aphis gossypii* and *Myzus persicae* (Homoptera: Aphididae) reared on potted chrysanthemums. *J. Econ. Entomol.* 85: 799–807.
- Xia, J. Y., W. van der Werf, and R. Rabbinge. 1999. Influence of temperature on bionomics of cotton aphid, *Aphis gossypii*, on cotton. *Entomol. Exp. Appl.* 90: 25–35.

Received 14 November 2005; accepted 19 March 2006.