Population Dynamics of Manure Inhabiting Arthropods Under an Integrated Pest Management (IPM) Program in New York Poultry Facilities—3 Case Studies

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Primary Audience: Flock Supervisors, Researchers, Extension Personnel, Plant Managers

Summary

Many arthropods inhabit caged-layer poultry manure, including pest and beneficial species. The primary pests are the house fly, *Musca domestica* L., and the darkling beetle, *Alphitobius diaperinus* (Panzer). The house fly is ubiquitous and rapidly infests newly accumulating poultry manure, often reaching enormous numbers. Flies pester farm workers, birds, and when emigration occurs, neighbors. The darkling beetle is a pest in older manure, causing damage to building infrastructure and if infested manure is spread onto fields, the beetles can fly in large numbers to nearby neighbors. The beneficial species comprise a mite and a beetle predator and a complex of parasitoid species, all of which can effectively reduce fly populations. In addition, the use of insecticides alone is no longer a viable fly management option. However, recent fly management developments that can easily be used by poultry producers now make a successful fly management program a reality. In this 20-wk study, arthropod populations under an intensive house fly integrated management program (IPM) in three high-rise, caged-layer poultry facilities in New York were monitored. Producers using IPM strategies successfully established the predatory hister beetle, *Carcinops pumilio* (Erichson), using three release strategies. Predatory mite populations were low throughout the study; however, strategic releases of parasitoids resulted in very high levels of house fly parasitism. Furthermore, the infrequent house fly outbreaks required only minimal insecticide applications. Producers using this IPM program successfully managed house fly populations in their facilities.

Key words: *Alphitobius diaperinus*, *Carcinops pumilio*, house fly, *Macrocheles muscaedomesticae*, parasitoid, population dynamics, poultry

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Description of Problem

House flies, *Musca domestica* L., are the primary pest of caged-layer poultry production in the Northeastern U.S. This insect continues to become more and more problematic as urban encroachment into traditionally agricultural areas increases. The use of insecticides has become increasingly unreliable due in large part to resistance development, whereas other insecticides have been removed from use through regulatory actions [1].

The major beetle pest infesting poultry manure is the darkling beetle, *Alphitobius diaperinus* (Panzer) [2]. This beetle can reach extremely deleterious numbers in caged-layer poultry facilities [3]. The larvae of this beetle move into building insulation and structural supports, seeking pupation sites, and in the process destroy the structure [4]. After manure has been spread on fields, adult beetles often fly to residential dwellings resulting in community-relations problems and lawsuits.

Integrated pest management (IPM) in modern poultry production includes a combination of cultural, biological, and chemical strategies used to lower pest densities to acceptable levels [2]. The first step in a poultry fly IPM program is to keep the manure as dry as possible, which reduces house fly production [5]. The primary indigenous house fly parasitoids found in New York poultry facilities are *Muscidifurax raptor* Girault and Sanders, *Nasonia vitripennis* Walker, and *Spalangia cameroni* Perkins [6, 7]. However, *Muscidifurax raptorellus* Kogen and Legner, a gregarious species, is commercially available and is currently being released in New York poultry facilities. The two most important fly predators are the hister beetle, *Carcinops pumilio* (Erichson) and the Macrochelid mite, *Macrocheles muscaedomesticae* (Scopoli) [8, 9]. Parasitoids can be purchased from commercial insectaries and released, whereas hister beetles can be collected and transferred from on-farm sources [10].

The succession of manure-inhabiting arthropods in poultry facilities has been examined [11, 12]. Indigenous house fly parasitoids, including *M. raptor*, *S. cameroni*, and *N. vitripennis*, have been shown to invade poultry manure in a newly built caged-layer facility within 8 wk after repopulation [11]. Small dung flies (Diptera:Sphaeroceridae) and acarid mites are the first arthropods to establish in manure in high-rise poultry facilities, followed by *M. muscaedomesticae* and hister beetles [12]. *Macrocheles muscaedomesticae* abundance reportedly peaked at 7 wk post cleanout, whereas hister beetle adults increased in number throughout the 16-wk cycle. When poultry manure is allowed to accumulate, predatory arthropods prosper, whereas weekly and monthly manure removal favors dipteran larvae [13].

This caged-layer poultry fly demonstration project incorporated our current knowledge of cultural, physical, biological, and chemical components into an IPM strategy aimed at minimizing the impact of house fly populations in poultry facilities. Our goal was to prevent the outbreak of adult house flies that is usually observed at 6 wk after repopulation. We also attempted to establish populations of the predatory hister beetle, *C. pumilio*, in these poultry facilities. Lastly, we observed the population dynamics of naturally occurring and augmented populations of arthropods commonly found in poultry manure in high-rise, caged-layer poultry facilities.

Materials and Methods

Study sites comprised three high-rise caged-layer poultry facilities, two located in Wayne Co., New York, and one in Erie Co., New York. The first facility, F1, was repopulated in mid-April, the second facility, F2, in early June, and the third facility, F3, in early July. Ventilation in F1 and F2 was positive-flow turbo in which air was pulled into the facility by fans located in the roof ridge and exhausted passively from the pit area. Manure dropping through narrow slots in the floor beneath the birds produced tall, sharply peaked manure mounds. The ventilation in F3 was of conventional design in which air was forced from the manure pit by pit-level exhaust fans, and manure fell directly to the pit floor, resulting in wide, somewhat flat manure mounds. Scraper boards were present in F1 and F2 and drop curtains were present in F3. Facilities F1 and F2 each contained 120,000 birds and F3 held 47,000 birds.

Producers followed IPM practices described below and included placement of manure drying

TABLE 1. Total and relative release levels of wild, captured, predaceous hister beetles and release levels of parasitoids into three New York caged-layer poultry facilities

FACILITY[A]	FACILITY AREA (m^2)	HISTER BEETLES (n)			PARASITOID COLONIES RELEASED/WEEK[B] (n)			
		Total released	Per bird	Density (m^2)	1, 2	3–5	6–8	9, 10
1	2,927	315,132	2.6	108	6	24	12	6
2	2,927	461,383	3.8	158	6	24	12	6
3	1,654	399,490	8.5	241	2.5	10	5	2.5

[A]Facilities 1 and 2 each held 120,000 birds; Facility 3 held 47,000 birds.
[B]Release rate during Weeks 1, 2, 9, and 10 was two parasitoids per bird; release rate during Weeks 3 to 5 was eight parasitoids per bird; release rate during Weeks 6 to 8 was four parasitoids per bird. Release rate was based on a mean of four parasitoids per pupa in release shipments.

fans, releases of predatory beetles and parasitoids, monitoring of house fly populations, and judicious use of insecticides. Due to high darkling beetle populations prior to cleanout, F3 was treated with cyfluthrin [14] 1 wk before repopulation. F1 and F2 were not treated for darkling beetles with a post cleanout insecticide.

Large fans (0.75 hp, 11,000 ft^3/min, 90 cm diameter) were suspended in the walkways of the manure pit, forcing air circulation along the manure rows, which dried the manure and aided in reduction of the fly population. These aisle fans were run constantly for 6 wk. F1 and F2 contained 16 fans, and F3 contained 8 fans. All fans were turned off after Week 6 of the study. Producers monitored their own manure pits daily for water leaks and feed spills as part of their standard operation procedures.

Weekly releases of commercially produced parasitoids [15] began 1 wk after the facility was repopulated. Shipments contained ca. 40% each of *M. raptorellus* and *M. raptor* and 10% each of *M. zaraptor* and *N. vitripennis*. The recommended release rate of house fly parasitoids in New York poultry facilities is four parasitoids per bird [10]. Thus, our release rate targeted an expected four parasitoids per pupa in commercial shipments. The distribution of parasitoids was varied intentionally to match the expected house fly availability. Therefore, during Weeks 1, 2, 9, and 10, one-half the normally recommended rate was released; during Weeks 6 to 8, four parasitoids per bird were released; and during Weeks 3 to 5, twice the recommended release rate was used (Table 1). Concentration of the high rate of parasitoid release into Weeks 3 to 5 was intended to help decrease the flush of house fly adults normally observed 6 wk after repopulation. It was expected that hister beetle populations would become established after 6 wk post cleanout and that additional parasitoid releases could be incrementally decreased. Weekly parasitoid shipments were distributed throughout the facility by sprinkling the parasitized fly puparia along the manure piles approximately one-third of the way up the manure pile.

Hister beetle adults were collected by the poultry producers from the manure pit prior to cleanout or were gathered from other facilities on the same farm. Beetles were released during Week 2 post cleanout in F1, during Week 4 post cleanout in F2, and daily from Weeks 1 through 7 post cleanout in F3. Producers were encouraged to collect as many beetles as possible with black light pitfall traps and Hister House traps (used according to the manufacturer's directions) [15] as previously described [16]. Beetles collected with pitfall traps were put through a sieve to remove darkling beetle larvae prior to transfer to the recipient facility. Contents from Hister House traps containing hister beetles were transferred to the recipient facility without sieving. The number of black light-collected beetles placed into each facility was estimated before beetle release by using subsample comparative weights. A subsample of Hister House traps was processed through Tullgren funnels to determine the estimated number of released beetles. All released beetles were scattered throughout the manure pit by farm personnel. Due to disease transmission concerns, only hister beetles collected from an individual farm were released on that farm. After the release period, no additional

hister beetles were introduced into the three facilities.

Sampling involved direct and indirect estimates of house flies, (adults and larvae), hister beetles (adults and larvae), parasitoids (adults), darkling beetles (adults and larvae), acarid prey mites, and predatory mites (*M. muscaedomesticae*). Spot cards and sticky cards were used to assess the number of adult house flies present in the manure pit weekly and semimonthly, respectively. Spot cards, 10 per manure pit, consisted of 76 × 127-mm (3 × 5-in.) white file cards placed on the manure pit walls and support beams, 1.5 m above the pit floor. Cards were replaced weekly, and the total number of fly spots and specks on one-half of each card were counted with a uniform grid. Sticky cards, 10 per manure pit, were single-sided, 76 × 127-mm cards [17] that were placed on a support beam ca. 1.5 m from the floor of the manure pit. Every second week, sticky cards were placed in the facility for 24 h and then removed, and the total number of house flies per card were recorded.

House fly parasitism rates were monitored semimonthly using sentinel house fly pupae [18]. Ten sentinel bags [8 × 8 cm (3 × 3 in), mesh density 5.5 squares/cm)], each containing 50 live house fly pupae, were placed using within block (manure row) randomly allocated locations on the surface, near the base of the manure pile. No two sites were used more than once. After 14 d, pupae were retrieved, and uneclosed pupae from each bag were held individually in size 00 gelatin capsules [19] in the laboratory for 8 wk to allow for fly and parasitoid emergence. The total numbers of uneclosed pupae, of successfully parasitized pupae, and of emerged parasitoids were recorded. Parasitoids were identified as to species, counted, and recorded. House fly parasitism was assessed at each sampling week by using two parameters: (1) total parasitism, which was the percentage of sentinel house fly pupae that were killed by parasitoid activity, and (2) successful parasitism, which was the percentage of sentinel pupae that produced adult parasitoids. Total parasitism was defined as successful parasitism plus all killed fly pupae that did not produce adult parasitoids; therefore, total parasitism must be higher than successful parasitism. Parasitism rates were corrected for control mortality [20].

All other arthropod populations present were assessed once every 2 wks with manure cores. Manure cores were removed with a bulb planter that collected ca. 400 mL of poultry manure [21]. Ten samples were collected from predetermined, random locations throughout each facility. All manure cores were taken from a location approximately one-half the distance from the top of the pile. Samples were placed in individual containers and held until placement in Tullgren funnels. Because of the large numbers of acarid prey mites present in samples, aliquot subsamples were removed, prey mites counted, and the number of prey mites per sample estimated. All other arthropod species were counted from the extracted samples.

The material, labor, and energy costs associated with administration of the IPM strategies used in this demonstration were calculated. These costs included operating the aisle fans, monitoring the facility, purchase and capture of beneficial insects, and applying insecticides.

RESULTS

FACILITY 1

The F1 was repopulated on April 20, 1998. A fly outbreak was observed in early June, and a pyrethrin fog was administered on June 5, 1998 (Figure 1). No additional insecticides were applied to this facility. House fly larvae peaked twice, during Weeks 4 and 10, whereas adult house flies captured on sticky cards peaked during Weeks 10 and 12 and then declined for the remainder of the study. The number of spots per card was high from Weeks 7 through 13. The facility manager was satisfied with the fly control at this location. In previous years, numerous insecticide applications were needed to manage house flies in this facility.

Predatory mite populations were not recovered until Week 8 and then declined throughout the study (Figure 2). Prey mites peaked during Week 10 and decreased rapidly. Successful parasitism rates reached 38% by Week 4 and 68% by Week 6 (Figure 2). This high level of successful parasitism remained throughout the study, dipping slightly during Week 10. Successful parasitism was primarily by *M. raptorellus* (58%), *N. vitripennis* (31%), and *M. raptor* (11%). Minor parasitism by *Pachycrepoideus vindemiae* (Ron-

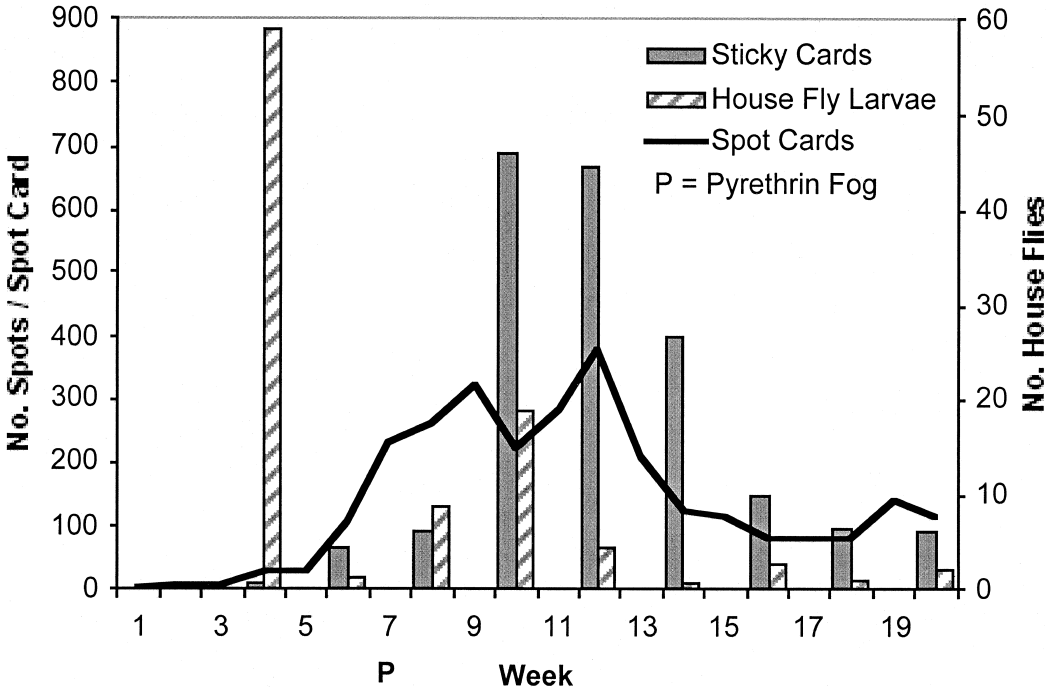

FIGURE 1. Dynamics of adult and larval house fly populations in Facility 1, a high-rise, caged-layer poultry facility in New York.

dani) (<1%) was also observed. Total parasitism closely mirrored successful parasitism until Week 12, when total parasitism approached 100% and remained there through completion of the study.

Hister beetle adults were released into F1 2 wk after repopulation. The total number of hister beetles released was estimated to be 315,132 with 266,172 captured using black lights and 48,960 captured with the Hister House (Table 1). Noticeable numbers of hister beetle adults and larvae were not recovered in Tullgren funnels until Week 8 followed by a large increase in numbers recovered during Week 10 (Figure 3). The numbers of adult hister beetles continued to increase through Week 16, whereas hister beetle larvae declined. Studies have documented that as larval darkling beetle densities increase, larval hister beetle numbers decrease [22]. Therefore, the decrease in hister beetle larvae at this facility might have been caused by increasing darkling beetle larval densities, decreasing prey mites and larval house fly populations, or both.

FACILITY 2

The F2 was repopulated on June 9, 1998. House fly populations were higher than those observed in F1 or F3 (Figure 4). Storm damage just after the facility was repopulated prevented installation and immediate operation of manure pit aisle fans. This delay, in combination with higher relative humidity in June, led to an apparently wetter manure pack than that observed in F1. In addition, this facility was newly constructed and, therefore, did not have any indigenous biological control agents. House fly larvae peaked during Week 4 (Figure 4). An outbreak of adult house flies occurred during Week 5 and again, as evidenced by sticky card sampling, during Week 9. The producer treated the facility with a pyrethrin fog on July 7, 9, and 16 (Weeks 5 to 7) and again on August 4.

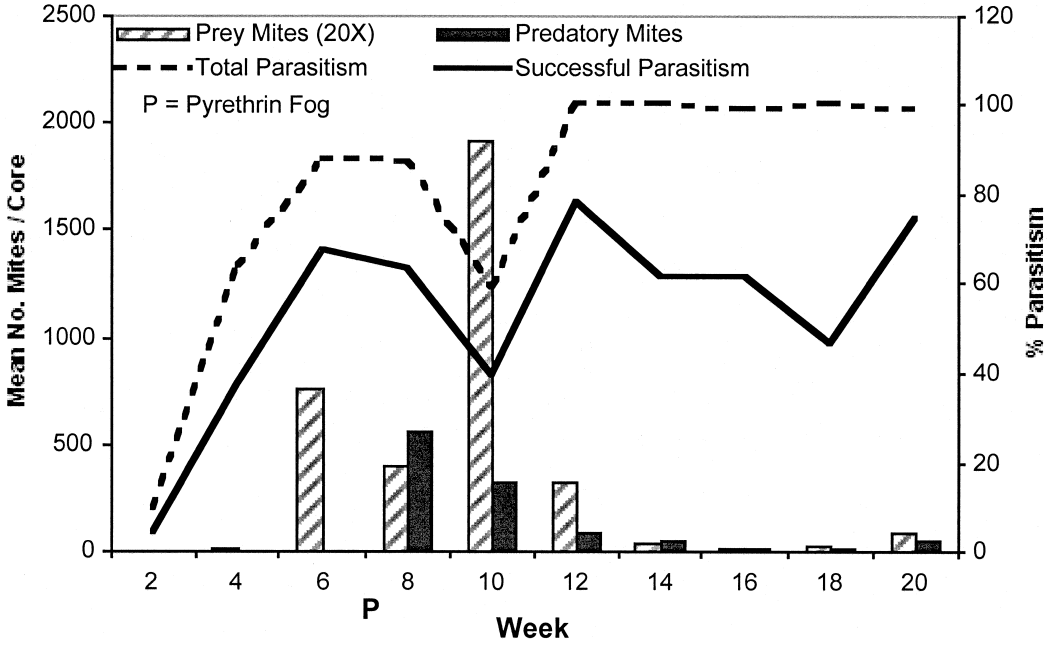

FIGURE 2. Predatory mites, prey mites, and percentage successful parasitism in Facility 1, a high-rise, caged-layer poultry facility in New York.

The first two insecticide applications were made with consultation, as the protocol directed; however, subsequent applications were made at the discretion of the producer. As estimated by spot cards, numbers of house flies were fairly stable after Week 7; however, the use of sticky cards revealed a fluctuating adult population (Figure 4). House fly larvae counted in manure samples presented a similar pattern to spot card data.

The farm manager was satisfied with the observed level of fly control at the conclusion of the study. The delay in manure pit aisle fan operation probably played a significant role in the house fly outbreak during Week 5. However, the number of insecticide applications was lower at F2 than applications made in previous years to other facilities on the same farm.

Infestation by mites was rapid, considering that this study was the first use of the facility with mites recovered from manure samples during Week 2 (Figure 5). However, prey mites peaked during Week 14 at about 21,000 mites per sample, one-half the level achieved in F1 but, nonetheless, remained above 10,000 mites per core for the remainder of the study. Predatory mite levels never exceeded an average of 230 mites per sample. Total and successful parasitism rates increased rapidly to 78 and 65% by Week 4; however, the rate declined to 29 and 13% during Week 6 (July 14, 1998), respectively (Figure 5). The decrease in parasitism is likely the result of the multiple pyrethrin applications in this facility. Total and successful parasitism rates rebounded to 98 and 80% in Week 8 before stabilizing around 100 and 50%, respectively, for the remainder of the study. Successful parasitism at F2 was primarily by *N. vitripennis* (45%), *M. raptorellus* (34%), and *M. zaraptor* (18%). Minor parasitism by *M. raptor* (<3%) and *P. vindemiae* (<1%) was also observed.

Adult hister beetles were released at F2, 4 wk after the birds were introduced. The total number of adult hister beetles released was estimated to be 461,383 with 424,183 captured using black lights and 37,200 captured with the Hister House (Table 1). Hister beetle numbers remained low until Week 12 when a sharp rise in numbers of adults and larvae were observed with a correspondingly low number of darkling

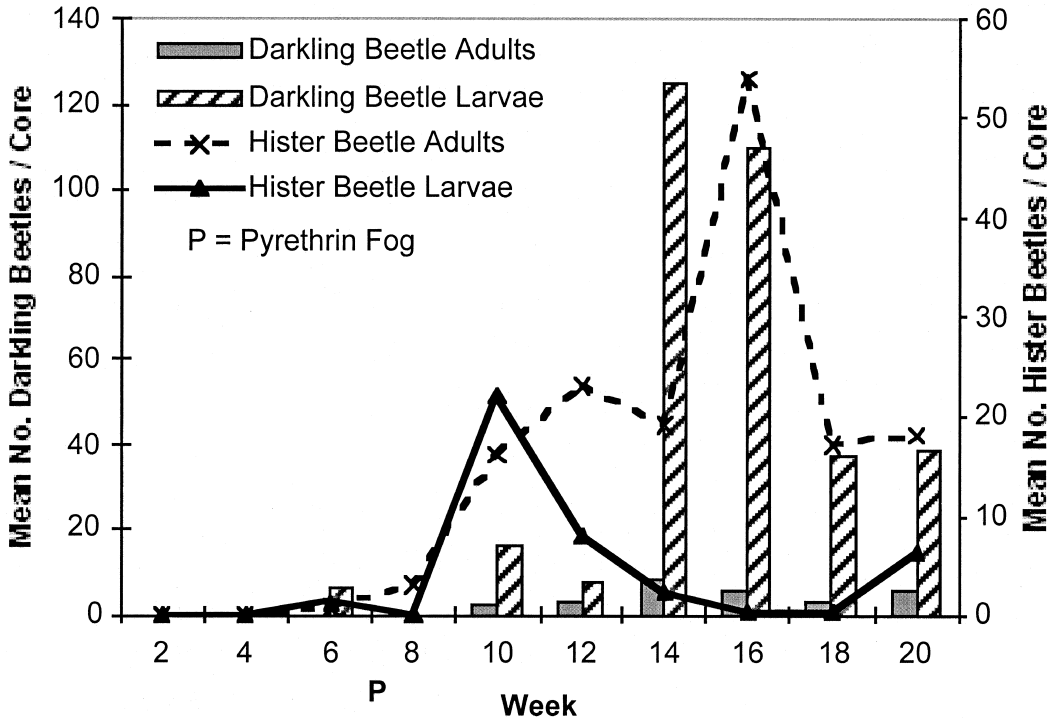

FIGURE 3. Hister beetle and darkling beetle population dynamics in Facility 1, a high-rise, caged-layer poultry facility in New York.

beetle larvae being recorded (Figure 6). Hister beetle numbers peaked during Week 16 and declined through completion of the study. As observed in F1 and F3, the numbers of larvae must be higher than the numbers of adults or population growth is limited. Hister beetles require about 40 d to develop from egg to adult, and the adult lives for about 100 d [23]; however, because of adult longevity, hister beetle populations can probably continue to increase while experiencing reduced adult recruitment (Figure 6). Because this facility was a new structure, it was not initially infested with darkling beetles. However, immature beetles appeared in manure samples from Week 6, and adults were present by Week 10, suggesting that darkling beetles infest a poultry facility rapidly.

Interestingly, similar to F1, the numbers of hister beetle larvae declined after a sharp rise in the darkling beetle larval population during Week 14. Increasing numbers of adult hister beetles were observed from Weeks 4 to 6, followed by a sharp decrease during Week 8. As was observed with the parasitoid data, the decrease in hister beetle populations happened soon after the pyrethrin applications, suggesting that multiple pyrethrin applications over a short period might have had a detrimental effect on the biological control agents in the facility (Figure 6). The pyrethrin applications probably killed the recently released adult parasitoids, thus reducing Week 6 parasitism. However, other studies have reported that hister beetles inhabit several centimeters of the manure pack, primarily near the peak [21]. Therefore, the effect was probably not manifested with the hister beetle population until multiple applications had been made and enough of the population had ventured to the surface, coming into contact with the insecticide.

FACILITY 3

The F3 was repopulated on July 2, 1998. The number of spots per card at F3 did not exceed 160, and the producer did not treat the facility with an insecticide for house fly control (Figure 7). In this facility, spot card, sticky card, and maggot counts all followed similar patterns.

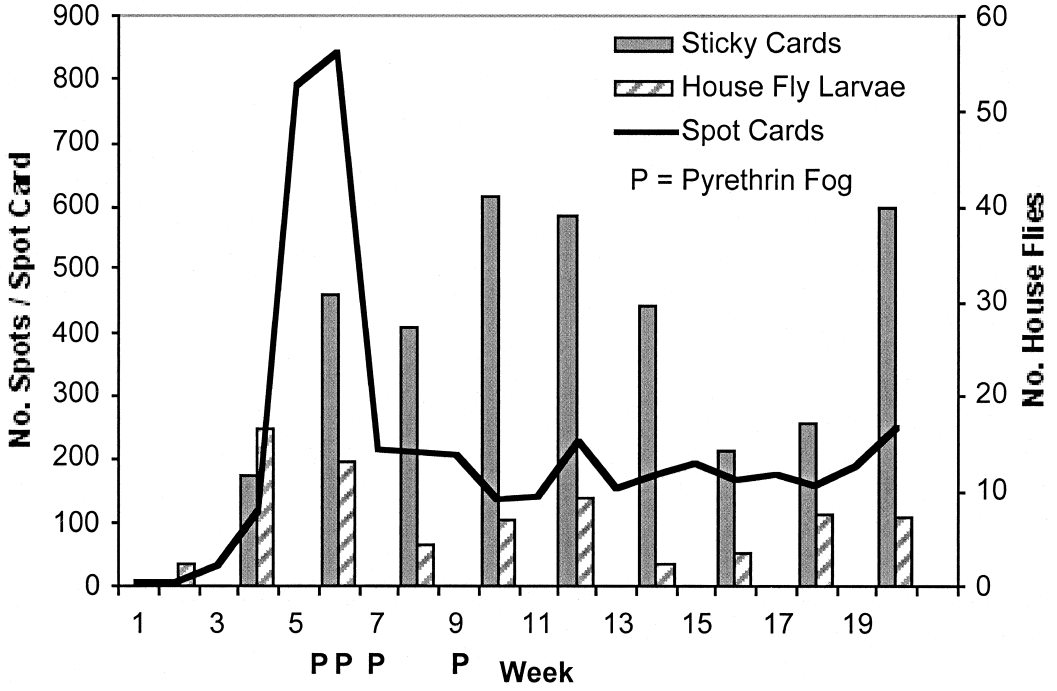

FIGURE 4. Dynamics of adult and larval house fly populations in Facility 2, a high-rise, caged-layer poultry facility in New York.

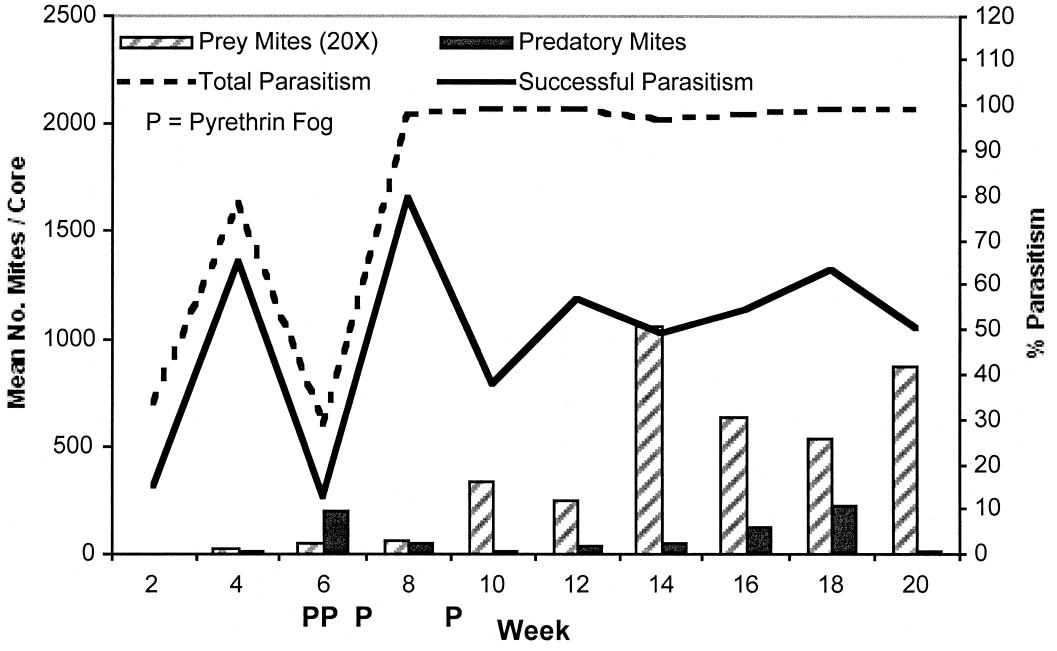

FIGURE 5. Predatory mites, prey mites, and percentage successful parasitism in Facility 2, a high-rise, caged-layer poultry facility in New York.

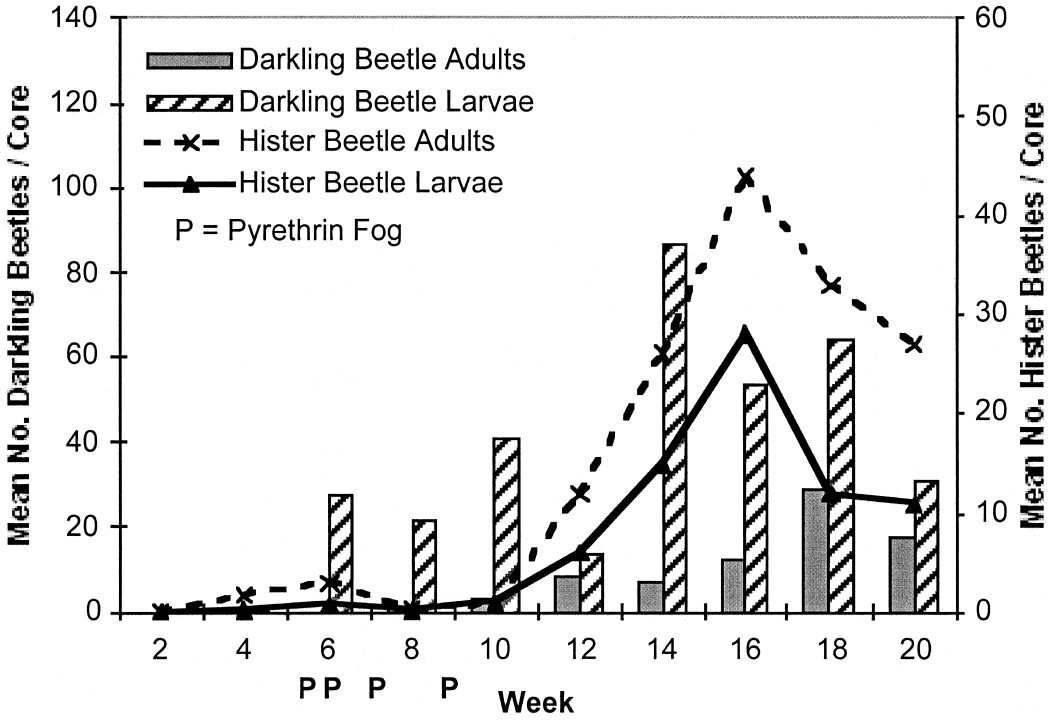

FIGURE 6. Hister beetle and darkling beetle population dynamics in Facility 2, a high-rise, caged-layer poultry facility in New York.

The farm manager of F3 was also satisfied with the level of fly control. This farm had received authorization to treat the facility with pyrethrins during Week 7; however, they decided to wait and the fly population subsequently decreased without an insecticide application. Fly control was excellent when sampling was terminated after Week 16.

Prey mites in F3 were abundant early in the study and increased through Week 10 (Figure 8). Predatory mites were not abundant, never exceeding 100 per sample. Total and successful parasitism rates did not surpass 40 and 12%, respectively, during the 14-wk sampling period (Figure 8). We attribute these rates to the absence of parasitoid recycling caused by a lack of fly pupae. The levels of parasitism that were observed reflect what producers generally observe after an initial release (parasitism resulting from the weekly releases). It is our opinion that the parasitoids were unable to successfully reproduce in this facility. Successful parasitism in F3 was primarily by *M. raptorellus* (59%), *M. raptor* (31%), and *M. zaraptor* (8%). A few specimens of *Spalangia* spp. and *N. vitripennis* were also recovered.

Hister beetles were released weekly in F3 beginning at Week 1 and continuing through Week 7. Hister beetle populations grew steadily during the study and were very high when the study was terminated (Figure 9). This facility housed only 47,000 chickens, compared to the 120,000 birds that were housed F1 and F2. The total number of adult hister beetles released into F3 was estimated to be 399,490 with all beetles captured by using black lights (Table 1). Captured beetles were released into the facility immediately after collection from an adjacent building. Based on the size of the facility, this release rate was much higher, on the magnitude of two- to threefold, than the release rates in the other two facilities.

Although hister beetles were released from Weeks 1 through 7, beetles were not recovered in samples in appreciable numbers until Week 8. After their appearance in the manure core samples, the populations of beetles increased rapidly, reaching a peak of about 55 and 51

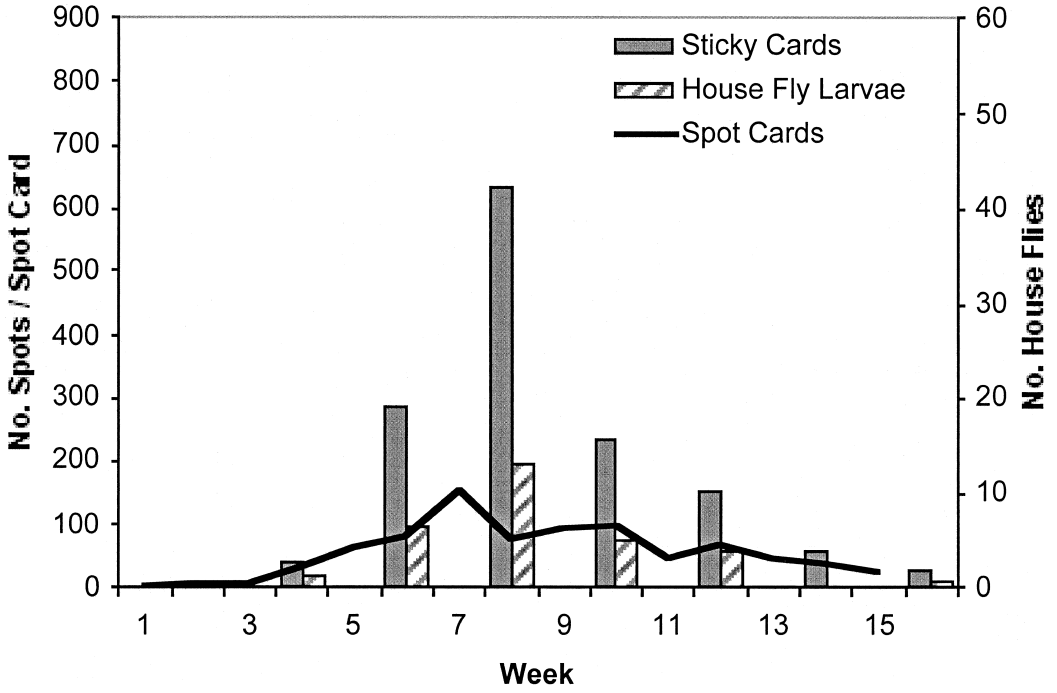

FIGURE 7. Dynamics of adult and larval house fly populations in Facility 3, a high-rise, caged-layer poultry facility in New York.

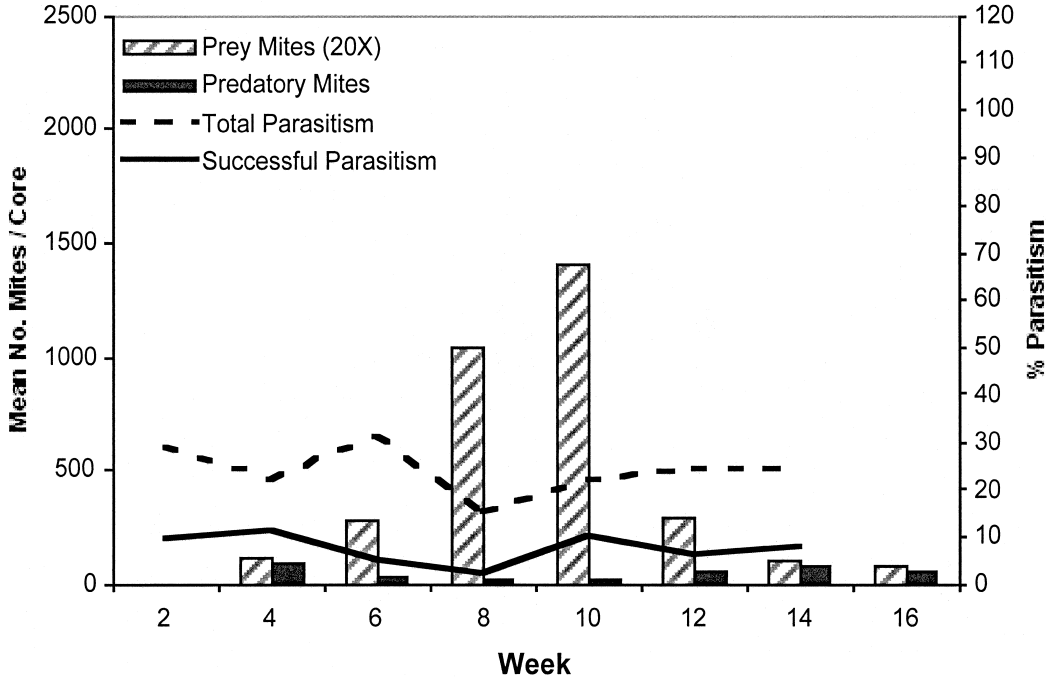

FIGURE 8. Predatory mites, prey mites, and percentage successful parasitism in Facility 3, a high-rise, caged-layer poultry facility in New York.

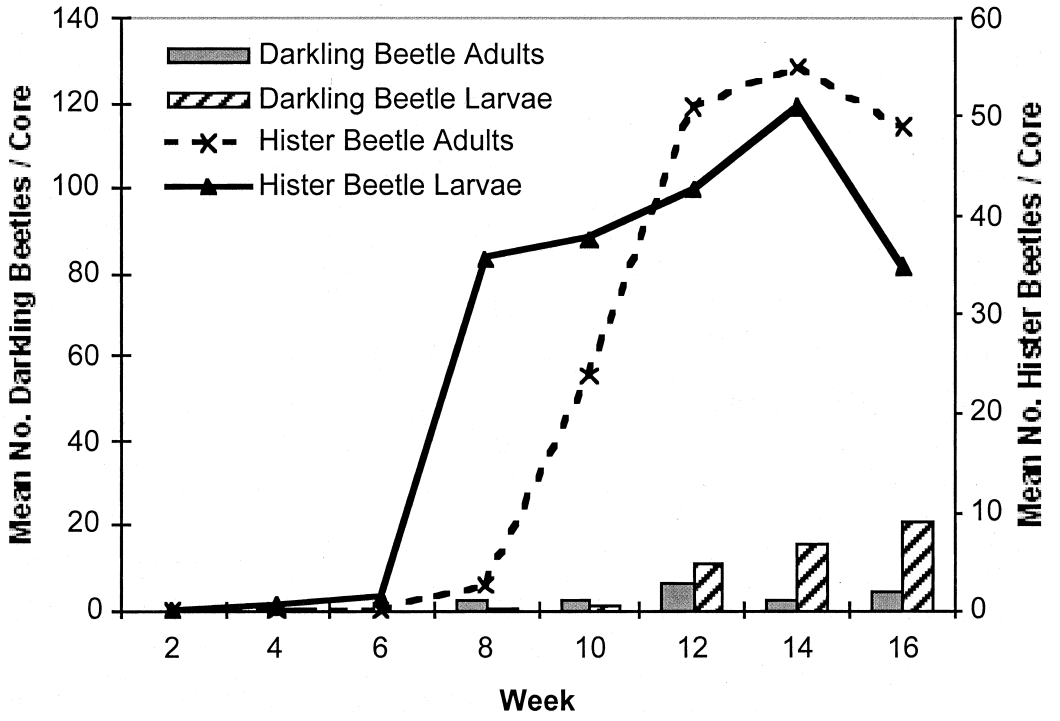

FIGURE 9. Hister beetle and darkling beetle population dynamics in Facility 3, a high-rise, caged-layer poultry facility in New York.

adults and larvae, respectively. These numbers were very high and represented an excellent fly management program. Because of the insecticide application in F3 prior to repopulation, the numbers of darkling beetles remained low throughout the study. This result was in contrast to the other two facilities and provided additional support to the theory that to attain high hister beetle populations, darkling beetle populations must be kept in check.

Discussion

In this study, we observed that increases in spot card numbers preceded, by 1 to 2 wk, large increases of house fly larvae collected from manure core samples, whereas elevated sticky card captures were indicative of concurrently high numbers of fly larvae (Figures 1, 4, and 7). This finding suggests that spot cards would be very useful in predicting an increase in fly larvae, thus providing producers with the opportunity to target an anticipated increase in fly pupae with increased parasitoid releases. Because sticky cards do not provide an advanced warning of increasing fly populations, their usefulness in a monitoring program appears to be quite limited.

Parasitoids were released in a staggered manner (two parasitoids per bird during Weeks 1, 2, 9, and 10; four parasitoids per bird during Weeks 6 to 8; and eight parasitoids per bird during Weeks 3 to 5), which resulted in a very high level of parasitism. This release strategy targeted the large numbers of house fly pupae that were expected to be present between Weeks 3 and 5. In facilities with larger larval house fly numbers, successful parasitism levels were very high (F1 = 58%; F2 = 62%), and total parasitism levels approached 100%, probably as a result of parasitoid recycling (in-field reproduction). Successful parasitism stabilized around 60%, probably due to the high ratio of recycled parasitoids to house fly pupae that resulted in increased parasitoid-induced mortality [24]. Parasitoid-induced mortality can result from several factors; however, repeated attacks on a house fly pupa by multiple parasitoids are a primary cause. In such a case, developing immature parasitoids

TABLE 2. Costs associated with administration of an integrated house fly management (IPM) program

		COST ($)		
STRATEGY	CATEGORY	Facility 1	Facility 2	Facility 3
Fans[A]	Electricity	1,350.00	900.00	483.00
Facility monitoring[B]	Labor	900.00	900.00	560.00
Hister beetle[C]	Black light	30.00	30.00	210.00
	Hister House	180.00	180.00	NA
Parasitoids[D]	Cost	1,451.00	1,451.00	643.00
Pesticides[E]	Chemicals	85.00	340.00	78.00
	Labor	15.00	45.00	80.00
Total cost		4,011.00	3,846.00	2,054.00

[A]Fans were 0.75 hp, 11,000 m^3/min, 90 cm diameter, run constantly for 6 wk. Facility 2 (F2) fans were in operation for 4 wk due to power outage. Fans were acquired from dismantled facilities on the farm and were recycled into their current use. Cost for new fans of similar size is $250.00 each.
[B]Labor associated with daily monitoring of the facility for water and feed leaks, fly monitoring, and other IPM tasks.
[C]Beetles were collected with black light traps for 2 wk in Facility 1 (F1) and F2 and for 6 wk in Facility 3 (F3). There were 90 Hister House traps used to collect beetles for F1 and F2. No Hister House traps were used to collect beetles in F3. Hister House traps cost $2.00 per trap.
[D]Parasitoids released for 10 wk. Cost = $10.50/colony plus $6.50 shipping per week. F1 and F2 received 132 colonies each, and F3 received 55 colonies.
[E]F1 received one application of pyrethrins + piperonyl butoxide (PBO), F2 received four pyrethrins + PBO applications, and F3 received one application of Tempo 20 WP.
[F]Facility 1 and Facility 2 each held 120,000 birds; Facility 3 held 47,000 birds.

are killed, and there is insufficient food for the newly laid parasitoid eggs to complete development. This scenario results in neither a house fly nor an adult parasitoid emerging from the pupa. Parasitoid recycling probably did not occur in F3 where fewer than 10 larvae per manure core were observed in samples from all but 1 wk.

Placing parasitized pupae in mesh bags and suspending these bags from rafters has been the standard method for parasitoid release. However, use of this method of release with *M. raptorellus* has two drawbacks: (1) low levels of parasitism (<7%) [25] and (2) minimal dispersal from the release site (<2 m) [26]. Our method of scattering the parasitized pupae on the manure surface removed this limitation and greatly improved parasitoid effectiveness. It appears that *M. raptorellus*, a relative newcomer to the complex of parasitoid species available to producers, can be a very effective component of a biological control program. This result is particularly evident when parasitized pupae are scattered on the manure and when the release rate is based on anticipated numbers of available fly pupae.

Pyrethrins are the insecticide of choice in IPM programs due to their effectiveness and lack of residual activity. However, the deleterious impact of multiple pyrethrin applications was observed in F2, in which high parasitism levels were observed during Week 4 (Figure 5). However, less than 15% parasitism was observed during Week 6 when three pyrethrin applications were made. Parasitism rebounded during Week 8 and declined during Week 10, when many of the parasitoids deposited during Weeks 6 and 7 would have likely emerged.

Poultry producers have a long history of using augmentative releases of parasitoids for fly control. Predator utilization in poultry pest management programs has been limited to natural colonization and conservation. However, the recent development of two trapping methods (black light pitfall traps and the Hister House) now allows producers to capture and release hister beetles into the manure pit, jump-starting the colonization process. In F1 and F3 where hister beetles were introduced earliest, beetles were not observed until Week 6, whereas in F2 adult hister beetles were recovered during the week in which they were released (Week 4). Dramatic increases in hister beetle populations were not observed until Weeks 10, 12, and 8 for F1, F2, and F3, respectively (Figures 3, 6, and 9). The delayed increase in hister beetles in F2 is likely the result of two factors: (1) multiple pyrethrin applications in that facility during Weeks 5 to 7 and (2) the increasing darkling

beetle larval population that prey on immature hister beetles.

High darkling beetle populations appeared to have a deleterious impact on hister beetle populations. At F1, hister beetle larvae were apparent and increased in the manure samples until darkling beetle larvae appeared (Figure 3). After an explosive increase in larval darkling beetles, the numbers of hister beetle larvae dramatically decreased. The number of darkling beetle larvae at F2 exceeded 20 per manure core by Week 6 (Figure 6). Correspondingly, the number of hister beetle larvae never exceeded the number of adults in manure samples. At F3, hister beetle larval numbers did not decline until Week 16 when larval darkling beetles exceeded 20 per manure core (Figure 9). Furthermore, hister beetle numbers were highest at F3 with 55 adults and 51 larvae recovered per manure core. These results, along with those of a previous study, provide strong evidence as to the extreme importance of monitoring darkling beetles and controlling them in caged-layer poultry facilities [22].

Producer costs associated with this demonstration are presented in Table 2. The purchase of parasitoids and electricity costs associated with running aisle drying fans were the greatest costs. However we did not take into account the value of the hister beetles that were added to the facilities. Currently, hister beetles are only commercially available as eggs and, because of their costs, are primarily used to establish populations in facilities that do not have existing hister beetles. There are no reliable estimates available for the value of adult hister beetles.

Producers operating the three poultry facilities in this study successfully established hister beetles under different release strategies, which suggests that predaceous hister beetles can be successfully established in poultry facilities with a variety of management strategies. High populations of darkling beetles at F1 and F2 probably impacted the hister beetle populations in these facilities. The house fly outbreaks at F1 and F3 were managed with one and no pyrethrin applications, respectively. The fly population at F2 required several pyrethrin applications. This requirement was the result of the nonfunctioning manure-drying fans and high humidity levels that resulted in wetter manure. Predatory mite populations were low throughout the study; however, strategic releases of parasitoids resulted in very high levels of house fly parasitism. Producers using this IPM model can successfully manage house fly populations in high-rise, caged-layer poultry facilities.

Conclusions and Applications

1. House flies were successfully managed in three caged-layer poultry facilities by using a IPM program that included innovative cultural control tactics, intensive biological control strategies with parasitoids and predaceous beetles, and judicious use of insecticides.
2. House fly parasitism reached and was maintained at very high levels in two of the facilities that were using a new targeted-release strategy.
3. Populations of the predaceous hister beetle were successfully established in poultry facilities using three different release methods.
4. High populations of darkling beetle deleteriously impacted hister beetle populations.

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