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Journal

Journal of Integrated Pest Management, 12(1)

ISSN

2155-7470

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Publication Date

2021

DOI

10.1093/jipm/pmaa023

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Journal of Integrated Pest Management, (2021) 12(1): 40; 1–10
doi: 10.1093/jipm/pmaa023
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Big Problems With Little House Fly (Diptera: Fanniidae)

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Subject Editor: Matthew Messenger

Received 23 September 2020; Editorial decision 4 November 2020

Abstract

The little house fly, *Fannia canicularis* (L.) (Diptera: Fanniidae), is a significant pest associated with livestock and animal systems worldwide. This species commonly develops in poultry production systems. The males of this species are a nuisance to people because they form mating swarms in enclosed spaces. The pest status of *F. canicularis* has not lessened since it was identified as a critical arthropod pest of veterinary importance over 50 yr ago. During this period, there has been little research progress to control this pest, especially when compared with other filth fly species. This article reviews the biology, distribution, pest status (including nuisance and pathogen transmission risk), monitoring, and control techniques, and identifies knowledge gaps for *F. canicularis*.

Key words: pest profile, review, lesser house fly, poultry, pest management

The genus *Fannia* includes a number of cosmopolitan and synanthropic pest species including *Fannia canicularis* (L.) (little or lesser house fly), *F. femoralis* (Stein) (coastal fly), *F. pusio* (Wiedemann) (chicken dung fly), and *F. scalaris* (F.) (latrine fly). These *Fannia* species develop in a variety of moist decaying organic substrates, especially animal feces, such as poultry feces, that contain relatively high amounts of nitrogen (Steve 1960, Greenberg 1985). Of the *Fannia* species, *F. canicularis* is the most widely distributed species and by far the most reported pest species. This article reviews the biology, economic impacts, and management of *F. canicularis* and discusses prominent knowledge gaps for which future research is needed.

Life Stages and Life Cycle

Adult flies in the family Fanniidae can be distinguished from other fly families by the presence of wings with a greatly shortened first anal vein (A1), with the second anal vein (A2) curving upward to cross an imaginary extension of the first anal vein, either at or before the wing margin (Fig. 1), and by having a bare or just slightly pubescent arista (Chillcott 1960, Rozkosny et al. 1997, Wang et al. 2007, Zhang et al. 2013). Adult *F. canicularis*, particularly females, can superficially resemble the house fly (*Musca domestica* L. (Diptera: Muscidae)), but are typically ~2/3 the body length (4–7 mm) of the house fly. They can also be distinguished from house flies by their less pronounced dark vertical thoracic stripes, by the way their wings are held directly over the back when at rest (Fig. 2; vs house fly's V-shaped wing orientation), and by lack of a strongly curved wing vein (R5) near the apex of the wing (Gerry 2015). As with many other flies, male *F. canicularis* have large holoptic eyes that nearly touch at the top of the head (Fig. 2), whereas females have dichoptic eyes that are well separated at the top of the head.

Eggs of Fanniidae are elongate, translucent white, and have dorsal wing-like longitudinal flanges (Fig. 3; Rozkošný et al. 1997). The role of these 'wings' is unclear though it has been suggested that they may allow the eggs to float (Lewallen 1954). *Fannia* species larvae look very different from the vermiform larvae ('maggots') typical of most other pest flies, including the house fly, which are characterized by having a smooth, cylindrical, and tapering body shape (Fig. 4). *Fannia* larvae are somewhat dorsoventrally flattened, have lateral extensions arranged in longitudinal rows, and possess two posterior spiracles on lobed stalks or tubes (Fig. 3). This is characteristic of the family (Rozkošný et al. 1997). Due to their smaller size and dark coloration caused by a highly sclerotized cuticle, *F. canicularis* larvae are well camouflaged in manure (Fig. 4). The larval habitats of *F. canicularis* are diverse and include animal feces, decaying plant material, compost, and even the nests of social Hymenoptera (Rozkošný et al. 1997). Immobile pupae closely resemble third instar larvae and encase the developing adult flies within (Fig. 3).

To complete development from oviposition to adult, *F. canicularis* require 22–36 d at 15–30°C (Lewallen 1954, Knoblock et al. 1977, Meyer and Mullens 1988), though development has been reported to be as fast as 18 d at 21–26°C (Thomsen and Hammer 1936, Tauber 1968), or as slow as 90 d at 10°C (Grzywacz 2019). Meyer and Mullens (1988) determined that total immature development time required approximately 572 degree-days above a developmental temperature threshold of 0.5°C. Eggs hatch within 32–48 h at ca. 26.5°C (Lewallen 1954, Steve 1960, Tauber 1968) or 48–72 h at 30°C

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(Grzywacz 2019), with the longer egg development time at the higher temperature giving an indication of the sensitivity of this species to high temperature (Meyer and Mullens 1988). Meyer and Mullens (1988) recorded egg hatch ranging from 2.5 to 6.1 d when held at 12–36°C. Egg development may occur in utero when females lack oviposition sites, resulting in egg hatch within minutes of egg deposition on a newly provided oviposition substrate (Anderson and Poorbaugh 1964). Egg hatch is greatly reduced at temperatures ≤ 4.5°C (<20% hatch) and eggs fail to hatch at temperatures ≥ 33°C (Grzywacz 2019), though Meyer and Mullens (1988) report 42% egg survival at 33°C and 12% egg survival at 36°C. Upon hatching, larvae exhibit phototaxis and move away from light (Brydon 1967). Larvae will also orient toward odors from nearby developmental substrate (Tauber 1968).

Fig. 1. The wing of *Fannia canicularis.* The second anal vein curves upward toward the first anal vein (arrow) (photograph by Stephanie Leon, UCR Entomology).

Meyer and Mullens (1988) report the mean development time for immature *F. canicularis* held at 15–30°C is 33–55, 46–74, 127–211, and 245–511 h for the first larval instar through the pupal stage, respectively. Tauber (1968) reports similar immature development times at 26.5°C with 36–60, 36–84, and up to 252 h for the first through third larval instar, respectively. Time to complete the pupal stage is unclear in Tauber (1968), perhaps as it is difficult to distinguish the pupal stage from the last larval stage as discussed above. Immature mortality is high at temperatures ≥ 33°C (Deal 1967, Meyer and Mullens 1988) and at temperatures < 4.5°C (Deal 1967).

Adults emerge from pupal cases primarily during daylight hours with timing of emergence entrained by light exposure patterns during immature development (Tauber 1968). Both males and females emerge concurrently and in an approximately 1:1 sex ratio (Tauber 1968), though earlier male emergence (protandry) is typical for other *Fannia* species (Tauber 1968, Zhang and Gerry 2015). General mating behavior of *F. conspicua* is discussed by Tauber (1968), with initial contact between the sexes occurring during flight. Adult females produce a pheromone (Z)-9-pentacosene that is slightly attractive to males (Axtell 1986). Mating occurs just prior to first oviposition (Tauber 1968). Only females over 96 h old (at 18.3°C and 66.6% RH) or 72 h old (at 23°C and 50% RH) will accept mating by males and lay fertile eggs, and only males over 48 h old can inseminate females (Tauber 1968). Time to first oviposition was reported by Lewallen (1954) and Steve (1960) to be 96–120 h at 26.6°C. Adult females require a protein food source to successfully mate and oviposit, resulting in no or very few eggs laid when female flies were offered a diet of only sugar and water (Tauber 1968). In the laboratory, males live up to 14 d and females more than

Fig. 2. Adult female (left) and male *Fannia canicularis.* The male eyes are larger and nearly touching at the apex of the head (photographs by Stephanie Leon, UCR Entomology).

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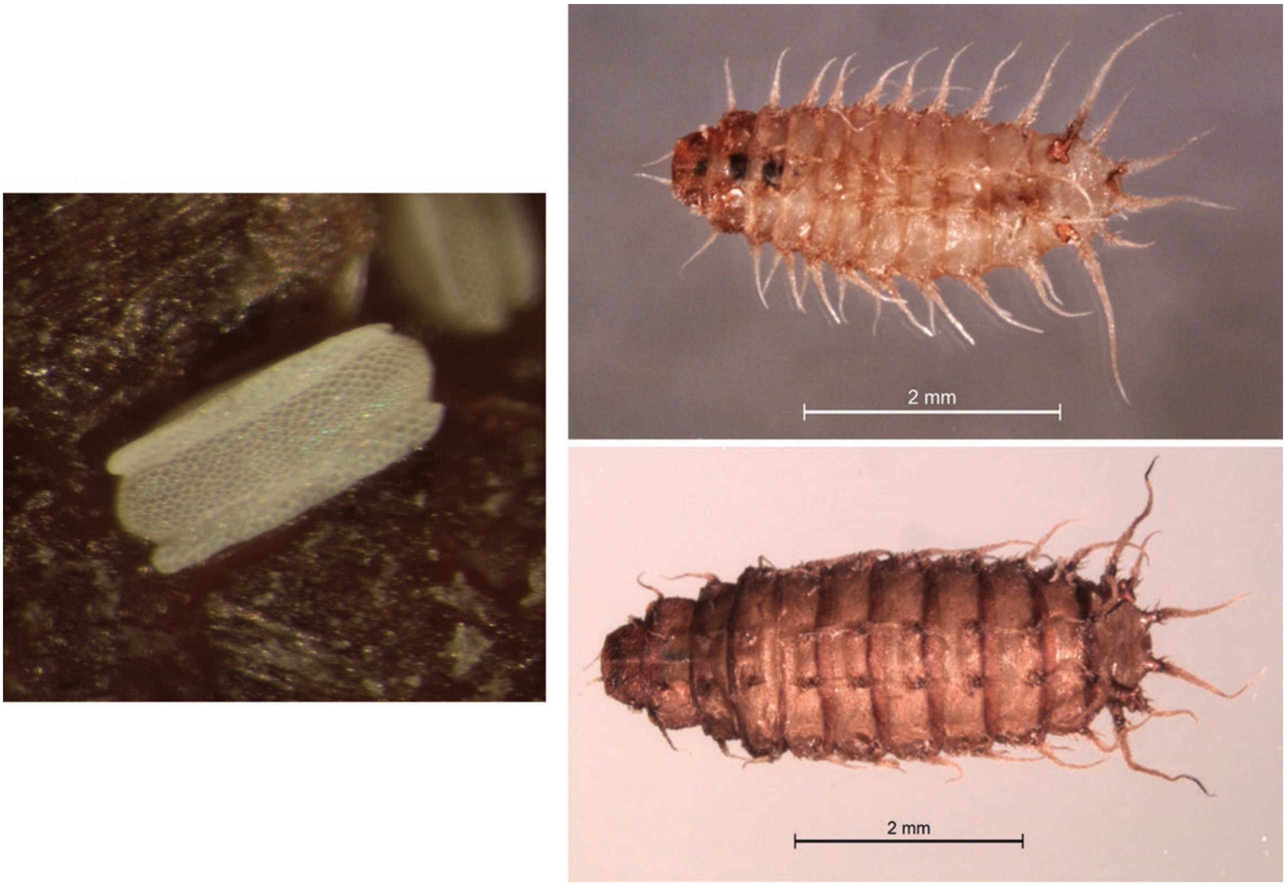

Fig. 3. Egg (left) of *Fannia canicularis* are translucent white with lateral, wing-like projections. Larvae (top right) are sclerotized with lateral projections. Pupae (bottom right) closely resemble the larvae (photographs by Alec Gerry [left] and Stephanie Leon, UCR Entomology [right]).

Fig. 4. Larvae of *Fannia canicularis* (solid arrows) in poultry manure are better camouflaged than house fly maggots (dashed arrows) due to differences in body color, body shape, and movement patterns (photograph by Caleb Hubbard).

24 d (Steve 1960). Typical adult life spans under field conditions are unknown.

Distribution

Fannia canicularis was spread by commerce and is now found worldwide (Chillcott 1960, Moon 2019), though an up-to-date distribution map is lacking. In North America, *F. canicularis* is found from Alaska to Mexico (James 1947, Chillcott 1960). They are generally distributed farther north than the house fly, being the dominant synanthropic fly species in many more northern locations such as Iceland (Deal 1967).

Adult *F. canicularis* are generally present from late fall through spring in the southern United States (Hewitt 1912, Hansens 1963), being most abundant in spring and disappearing in summer in geographic areas where summer temperatures are high (Lewallen 1954). In coastal southern California, *F. canicularis* (and *F. femoralis*) are most abundant in spring through early summer (February–June) before being replaced by the house fly during the hot summer months (Brydon et al. 1966, Legner and Brydon 1966). Where winter temperatures are mild, *F. canicularis* will overwinter as larvae or pupae, becoming active again in spring as temperatures increase (Sychevskaia 1954). Adult *F. canicularis* have lower and upper temperature thresholds for activity of 4.5 and 40.8°C, respectively (Nieschulz 1935), indicating less tolerance to high temperature relative to *M. domestica*, which has a lower and upper temperature threshold of 6.7 and 46.5°C, respectively (Lewallen 1954). When daily high temperatures exceed 32°C, *F.* canicularis are generally replaced by the house fly as the main pest in animal production systems where *F. canicularis* are common (Deal 1967).

Females lay eggs in moist organic substrates (especially poultry feces) with 55–65% moisture being preferred, though females will oviposit on substrates with moisture content as low as 45% (Mullens et al. 2002) and immatures can complete development in substrates with moisture content as low as 30% (Fatchurochim et al. 1989,

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Mullens et al. 2002). This range may contribute to the ability of *F. canicularis* to exploit a wide range of developmental substrates (e.g., Rozkošný et al. 1997). In contrast, house fly immatures prefer substrates with 50–75% moisture content and are less tolerant of substrates with low moisture content (Fatchurochim et al. 1989).

Pest Status and Damage

Animal Production

Fannia canicularis is often a pest in poultry facilities where feces are allowed to accumulate, undisturbed by both birds and human facility managers. Feces regularly build up beneath birds in suspended wire cages or over slatted flooring (Steve 1960, Axtell 1985). This is especially true in intensive poultry production systems where birds are excluded from contact with their feces, such as breeder and egg-layer poultry production (Machtinger et al. 2020). Breeder birds are generally housed in facilities with nesting boxes and feeders placed on raised wooden slat floors that allow bird feces to fall through and accumulate beneath the slats where they cannot be disturbed by the birds. Egg-layer birds are often held in suspended wire cage systems, with feces and spilled feed accumulating in rows beneath the wire cages.

Turkeys and broiler chickens are generally housed in wide single-story buildings where birds can move freely across a floor covered with wood shavings or other bedding material (Axtell 1985). In these houses, birds disturb the feces and mix it into the bedding so that it does not accumulate to provide a development site for immature flies. Disturbing the feces and bedding material also encourages drying of the manure to reduce oviposition opportunity for flies.

Fannia canicularis is also reported to develop in the feces of other animals, including swine, horses, cattle, sheep, and humans (Ogata et al. 1957, Steve 1960, Greenberg 1985). Additionally, they can be a serious pest in fur mink production (Bland 1964, Funder and Mourier 1965), although mink feces alone are reported to be a poor developmental substrate for this fly (Bland 1964, Saha 2018). In addition to development in animal feces, *F. canicularis* has been reported to develop in pickling tubs in Japan (Ogata et al. 1957), compost and rubbish (Ogata et al. 1957), outdoor lavatories (Ogata et al. 1957), and on clothing or body coverings contaminated with feces or urine, and in decaying animal flesh (Benecke and Lessig 2001, Bonacci et al. 2017).

Nuisance

Fannia canicularis adults are diurnally active and widely dispersed on poultry ranches during the day (Anderson and Poorbaugh 1964). Adult flies can disperse to nearby residential homes where they can pose a nuisance to homeowners (Meyer et al. 1987, Meyer 1993). *Fannia canicularis* often enter homes and/or form mating swarms in areas protected from wind or direct sunlight, such as beneath trees and shade structures, or in covered porch areas (Zeil 1986, Rozkošný et al. 1997). Females aggregate on walls and ceilings within poultry houses as well as in trees and shrubs outdoors (Lewallen 1954, Anderson and Poorbaugh 1964). Males typically fly in tight circles about 2 m above the ground, placing them about face height for humans. Steve (1960) noted, 'At times, adult playflights contained such large numbers of flies and became so dense as to force persons to close their eyes and mouths while walking through them'. The sex ratio of flies within a mating swarm is 12:1 M:F (Bland 1964), so it is the male flies that are often responsible for the nuisance reported to local health departments. Near poultry farms in California, it has been suggested that *F. canicularis* is a more important nuisance pest than the house fly, as fly complaints aligned with seasonal peaks in *F. canicularis* activity (Meyer 1993). Fly nuisance can result in citations issued to the animal facility as well as additional costs related to fines or emergency fly management (Anonymous 1955, Meyer 1993). Currently, there are no methods available to reduce the movement or dispersal of *F. canicularis* from development sites on a poultry facility to neighboring properties, but increased air movement using fans at neighboring properties is suggested to disrupt mating swarms and reduce nuisance impacts (Bland 1964, Gerry 2015).

Disease Transmission

Because *F. canicularis* can develop in animal feces and adult flies will also contact feces to oviposit or feed, they are potential vectors for several important animal and human pathogens. Viruses include virulent Newcastle disease virus (Rogoff et al. 1975, Chakrabarti et al. 2007) and Aleutian mink disease virus (Prieto et al. 2018). Bacteria include *Campylobacter* spp. (Royden et al. 2016), *Bacillus subtilis*, *Enterococcus* spp., *Staphylococcus aureus*, and antimicrobial-resistant strains of *Pantoea* spp. (Boiocchi et al. 2019). *Fannia canicularis* and *F. scalaris* are intermediate hosts of an eye worm (*Thelazia californiensis* Price; Burnett et al. 1957, Steve 1960).

Fannia canicularis and *F. scalaris* have been associated with rare cases of ear, urinary tract, intestinal, or auricular myiasis in humans (Hewitt 1912, James 1947). Cases result from host contact with clothing soiled with feces containing fly eggs or larvae, or from accidental consumption of fly eggs or larvae in contaminated food (Chillcott 1960, Rozkosny et al. 1997, Benecke and Lessig 2001, Bonacci et al. 2017).

Fly Management (Integrated Fly Management)

The integrated pest management (IPM) principle (Stern et al. 1959, Flint and Van Den Bosch 1981) is rooted in the foundation of linking pest density to economic damage and costs of control. In the case of a nuisance fly such as *F. canicularis*, this threshold can be very subjective. Meyer (1993) noted that, 'the "tolerance threshold" (the fly density which stimulates homeowner action) declined as urbanization continued to increase'. Although it may be difficult to declare a specific action threshold for *F. canicularis*, monitoring fly activity is still key to developing an on-farm IPM program.

Monitoring

Sticky fly ribbons deployed for defined intervals can be utilized to successfully collect adult *F. canicularis* for identification and to monitor changes in fly abundance or activity, in terms of catch rates (Steve 1960). Bram et al. (1974) suggested recording visual counts of swarming male *F. canicularis* to determine change in fly activity over time. It was suggested that at each poultry house, five different male swarm locations are identified and the number of swarming flies within a 5-foot-diameter field of view at each location is counted. It was even suggested that ≥20 flies per swarm was an indication that fly activity was too high and treatment to control adult flies should be initiated. Anderson and Poorbaugh (1964) reported that both sticky ribbons and visual fly counts showed a similar fluctuation in *F. canicularis* activity at a poultry facility, but the use of sticky ribbons is recommended as visual counts may vary by experience of the person conducting the activity count.

Sticky ribbons are best placed near roof level within animal housing because *F. canicularis* congregate at this location from mid-morning to late afternoon (Anderson and Poorbaugh 1964), and the ribbon serves as a landmark that flies will approach and land on

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Zeil (1986). In California, sticky fly ribbons have also proven useful for monitoring the activity of *F. femoralis* at poultry facilities (A.C. Gerry, unpublished data) and will capture other pest fly species as well, making this method a suitable method for monitoring each of these pest fly species simultaneously (Gerry 2020). Sticky ribbons should be checked often as the trapping surface can become compromised by dirt, humidity, or covered in flies in less than 24 h.

Monitoring *F. canicularis* activity can also be achieved using 'spot cards', which are 3 × 5 in (7.6 × 12.7 cm) white index cards placed at locations where these flies are noted to land and deposit regurgitation and fecal spots (Axtell 1970). These fly spots may be used to monitor relative adult populations over time, though similar spotting by other fly species can limit the effectiveness of this monitoring approach when they are abundant. When multiple pest fly species are present, spot counts provide a measure of overall fly activity (Lysyk and Axtell 1986). Spot cards can be used to monitor the activity of specific fly species during times of the year when only a single species is active. For example, Lysyk and Axtell (1986) suggest that spot cards can be used to monitor *F. canicularis* activity early in the season when this species constitutes a large proportion of the fly population at many poultry facilities. In contrast to fly tapes, it is recommended to avoid placing spot cards near the ceiling of open California-style poultry houses since high daytime temperature reduces fly activity during the day at this location (Lysyk and Axtell 1985). The cards should be left in place for a period of time (often a week), then the resulting deposition spots can be counted as a measure of fly activity during the sampling period. Simultaneous use of sticky fly ribbons to measure fly diversity will support interpretation of spot card counts. An increase or decrease in spot numbers over time will provide insight into fly population abundance, with the absence of spots indicating flies are not a problem during a given week.

Immature *F. canicularis* can be sampled from manure using a core sampler inserted horizontally into the manure cone (Eastwood and Schoenburg 1966). The core sample is then placed into a Berlese funnel to collect larvae (Brydon and Fuller 1966), washed through a sieve to retain immature flies (Eastwood and Schoenburg 1966), or placed into water with a high salt concentration to float immatures (Southwood 1978). Using a Berlese funnel, all *F. canicularis* larvae that can be captured from the sample will be collected within a period of 18 h when using a 100-W incandescent light bulb (Brydon and Fuller 1966). Emergence traps are another useful tool for assessing *F. canicularis* abundance, and may be related to future adult fly abundance (Cook et al. 1999). Core samples can be placed into emergence traps, or emergence traps can be placed directly onto substrates where flies are thought to develop and emerge from. Allowing adults to emerge is advantageous because adult flies are often easier to identify than immature flies (as key characteristics are more easily distinguished). Emergence trap counts are probably a better measure of future pest status of the adult fly population relative to other immature sampling methods as emergence traps record adult flies that successfully complete development.

Immature fly density will vary considerably among small substrate samples. To address this variability, it is appropriate to pool small substrate samples from a substrate source into a single larger volume of substrate for which immature abundance is recorded. The total number of sample pools that should be acquired will depend on the desired level of precision in the mean density count, with percent deviation from the mean count calculated after processing each sample pool until the predetermined level of precision is met (Schoenburg and Little 1966).

Mechanical Control

Manure and moisture management have historically formed the basis of *F. canicularis* population suppression in poultry production facilities; however, even in open-sided 'California style' egg-layer houses, airflow and high temperature may be inadequate to dry manure sufficiently to limit *F. canicularis* breeding (Axtell 1970). In open-sided 'California-style' layer houses, density of *F. canicularis* and *F. femoralis* was reduced when poultry feces deposits exceeded 12 inches in depth resulting in more rapid drying of feces coupled with increasing numbers of fly predators and parasitoids as a result of greater habitat stability (Legner et al. 1973).

Regular disturbance of poultry feces either by birds with access to feces or by human actions can provide a substantial level of fly control as this encourages rapid drying of manure. In some situations, accumulated manure cannot be dried sufficiently by human disturbance and fly production will continue (Stone and Brydon 1965). Where feces are not so disturbed or disturbance is insufficient to reduce immature fly density, fly production can be suppressed by 'frequent cleanout' of poultry houses where poultry feces are removed fully or in part from poultry houses each week. To prevent continued larval development in manure removed from the poultry house, this material should be rapidly and thoroughly dried or suitably composted with pile turning to enhance heat cycles (Eastwood et al. 1967, Cook et al. 1999, Gerry et al. 2005). If composting manure, addition of bulking agents (carbon source) will increase compost pile temperatures, resulting in the elimination of *F. canicularis* larvae within 3 d and near elimination of house fly larvae within 4 d, although the subsequent cooling of the pile will allow the return of immature house flies within 8 d if the pile is not turned (Eastwood et al. 1967). Because composting also kills pathogens in the feces (e.g., Hess et al. 2004), composting is the preferred method for handling poultry feces removed from a poultry house prior to final disposal.

Complete removal of poultry feces and litter from a poultry house may disrupt beneficial insect populations, while some *F. canicularis* larvae may remain to complete development in feces residue left behind in depressions or near walls and structural supports (Peck and Anderson 1970). Furthermore, due to short life cycles and high fecundity, filth fly populations rebound more quickly than do populations of fly predators and parasitoids. To conserve natural enemies and thereby limit fly resurgence, alternatives to full manure removal from poultry houses have been examined (Legner et al. 1966, 1973; Peck and Anderson 1970; Legner and Olten 1971; Mullens et al. 2002). Partial cleanouts (allowing some portion of the manure to remain) could conserve a reservoir for predators and parasites, and alternate-row cleanouts are suggested to hasten recolonization of new manure piles from adjacent, undisturbed manure (Hinton and Moon 2003). However, in one study alternate-row manure removal failed to control pest flies, and *F. canicularis* populations actually increased after disturbing manure (Mullens et al. 1996). Peck and Anderson (1970) found that monthly cleanouts led to the greatest resurgence of *Fannia* species, with longer cleanout intervals required for predators to recover. Most of this work was conducted in California, and results may vary by region.

Targeting adult flies using attractant-baited traps may help to reduce nuisance fly populations. Traps baited with acetic acid or ethanol alone or in combination trapped large numbers of *F. canicularis* (Hwang et al. 1978, Landolt et al. 2015). Like many insects, *F. canicularis* are attracted to black lights, and the use of ultraviolet (UV) traps may reduce adult numbers in closed spaces (Bland 1964, Tarry 1968). There are no known adverse effects on egg production or weight gains resulting from continued use of UV light traps in poultry houses (Hogsette et al. 1997, Hogsette and Wilson 1999). Currently, there are no methods available to reduce the movement or dispersal of adult *F. canicularis* from development

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sites on a poultry facility to neighboring properties, but increased air movement using fans at neighboring properties is suggested to disrupt mating swarms and reduce nuisance impacts (Bland 1964, Gerry 2015).

Biological Control

Like other flies, *F. canicularis* is susceptible to a range of parasitoids, predators, and pathogens (Steve 1959). Opportunistic predators such as spiders, gamasid mites, scatophagid flies, and carpenter ants will prey on immature and adult *F. canicularis* and *F. femoralis* (Steve 1959). When *Fannia* species occur in the same manure habitat as the house fly, predatory histerid beetles will prey on both fly groups (Rezende et al. 2018). The entomopathogenic fungus *Entomophthora muscae* (Cohn) Fresenius can infect *F. canicularis*, with an average infection rate of 15% and a maximum rate of 40–45% in southern California (Steve 1959, Mullens et al. 1987). Male *F. canicularis* are more likely to be infected than females.

Although few *Fannia*-specific parasitoids have been identified, several parasitoids of other filth flies will also exploit *Fannia* species (Legner et al. 1966, Legner et al. 1967). On coastal southern California poultry facilities, hymenopterous parasitoids recovered from *F. canicularis* include the ichneumonid wasp, *Stilpnus anthomyidiperda* (Viereck), and the pteromalids *Muscidifurax raptor* Girault and Sanders (Hymenoptera: Ichneumonidae) and *Spalangia endius* Walker (Hymenoptera: Pteromalidae) (Legner et al. 1966, 1967; Legner and Olten 1971). The same pteromalids parasitize the coastal fly, *F. femoralis*, with *M. raptor* and *S. endius* dominating during periods of cool and warm weather, respectively (Legner and Brydon 1966). In Massachusetts, another pteromalid, *Pachycrepoideus vindemmiae* Rondani, was recovered from both *F. canicularis* and *F. scalaris* (Steve 1959). Although entomopathogenic nematodes commonly attack other filth fly species, no infections of *F. canicularis* by *Steinernema feltiae* Filipjev or *Heterorhabditis heliothidis* (Khan, Brooks & Hirschmann) were detected in poultry manure, perhaps due to inhospitable aspects of the manure (Mullens et al. 1987).

Chemical Control

Larvicides and Manure Treatments

Insecticides may be applied by spray directly to poultry feces to kill exposed immature and adult flies, though constant deposition of fresh feces simultaneously dilutes and buries the active ingredients. Application of insecticides or insect growth regulators (IGRs) may still be useful as a spot treatment to control immature flies where poultry feces are wet due to water leaks or diarrhea. Regular pesticide applications can be made to manure immediately after stirring the manure to expose immatures (Stone and Brydon 1965). Application of a mixture of diazinon (an organophosphate insecticide) and gypsum to stirred manure provided good control of *F. canicularis* when applied at approximately weekly intervals (Stone and Brydon 1965).

A better approach is to apply insecticides that specifically target the developing immature flies within the poultry feces. Cyromazine is an IGR that selectively disrupts fly larval–pupal metamorphosis with little or no impact on beneficial fly predators (Axtell and Edwards 1983, Meyer et al. 1987). Cyromazine can be applied as a 'feed-through' insecticide (Larvadex), with the IGR mixed into poultry feed. Cyromazine is effective against early instars of *F. canicularis* when applied as a feed-through at an adequate rate (5 ppm; Mulla and Axelrod 1983, Meyer et al. 1987). When provided in poultry feed at 5 ppm, cyromazine reduced development of *F. canicularis* and *F. femoralis* at temperatures from 15.5 to 27°C, but when applied at a lower concentration of 1.5 ppm, development was reduced only at 21–27°C (Meyer et al. 1987). Providing poultry with feed mixed with cyromazine continuously was more effective than providing cyromazine in feed in alternate weeks, with immature *F. canicularis* and *F. femoralis* reduced within one week of IGR delivery followed by reduced adult populations 4 wk later (Meyer et al. 1987). In some U.S. states, cyromazine is not registered for use as a feed-through and must be used as a water-soluble granule (Neporex 2SG, 2% cyromazine) applied directly to poultry feces. Application of Neporex 2SG to accumulated poultry feces in an egg-layer facility provided good control of *F. canicularis* (and house fly) for up to 7 d when broadcast as a dry granule and up to 21 d when dissolved in water and applied as a spray (Donahue et al. 2017).

Chemicals that alter the pH or other chemical properties of animal feces can reduce suitability of the feces for fly development. First-instar *F. canicularis* appear to be more susceptible to changes in pH relative to older instars as first-instar *F. canicularis* were found in poultry feces only within a narrow pH range (7.25–9.24), while later instars were often found outside this pH range (Brydon et al. 1966). Sodium bisulfate is a pH reducing chemical that, when added to animal feces, can limit bacterial growth, alter bacterial species diversity, and reduce adult fly emergence (Terzich et al. 1998, Sweeney et al. 2000, Calvo et al. 2010). Although most studies using pH reducing compounds (such as boric acid, acetic acid, borax, or calcium cyanamide) were focused on management of house fly (Lachance et al. 2016, Cook et al. 2018), it is likely that pH manipulation would impair development of *F. canicularis* and other filth flies that consume bacteria during their immature development; further investigations are warranted.

Adulticides

At poultry facilities with California-style houses, *F. canicularis* are encountered outdoors during the day but will aggregate in indoor ceiling areas at night (Anderson and Poorbaugh 1964, Anderson 1965). Targeting this indoor ceiling area of adult fly aggregation for application of insecticidal materials can achieve control with the least amount of insecticide (Anderson 1966). In Oregon mink farms, the organophosphate insecticides ronnel and dimethoate (both applied at 1% concentration) produced rapid knockdown of *F. canicularis* with residual efficacy of both insecticides lasting at least two weeks (Bland 1966). In controlled studies, flies were well managed by early season manure removal followed by application of insecticides, with *F. canicularis* more readily controlled than the house fly (Axtell 1970).

Because of the predilection of adult flies for resting on lines, insecticide-treated vertical cords placed at indoor locations have proven useful to control *F. canicularis* (Ogden and Kilpatrick 1958, Steve 1960, Williams 1973). Insecticide-treated cords are most effective when stretched tightly and placed out of drafts (Williams 1973).

It is well known that repeated use of the same active ingredient or class of insecticide can lead to insecticide resistance and elimination of beneficial predators. Insecticide resistance in *F. canicularis* has been documented (Meyer et al. 1990), but most of the exposure was considered to be secondary to applications made for house fly control (Georghiou et al. 1967). Resistance to malathion was already evident by the 1960s (Bland 1964), likely due to overuse of this chemical for house fly control.

Insecticidal Baits

Dry granular baits formulated for house flies provide little control of *F. canicularis* (Ogden and Kilpatrick 1958). Although

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F. canicularis is susceptible to the active ingredients in current fly baits (Murillo et al. 2015), *F. canicularis* will not readily feed on these baits when scattered on the ground as per the label. No efforts have been made to optimize current fly baits or design new baits to attract and encourage feeding by *F. canicularis*.

Knowledge Gaps and Needs Assessment

Twenty years ago, key knowledge gaps concerning the biology and ecology of *F. canicularis* were identified by veterinary entomologists (Geden and Hogsette 2001, Machtinger et al., 2020). Since then, the pest status of *F. canicularis* has not lessened, yet little research progress has been made, especially when compared with other filth fly species (e.g., Geden et al. 2020). A review of the literature suggests that a 'golden age' for *F. canicularis* research existed in the 1960s, which was followed by a sharp decline in research effort (Fig. 5). A recent uptick in publications suggests that *F. canicularis* is as problematic as ever. Many of these recent publications are surveys of nuisance and/or synanthropic flies from outside of the United States, highlighting the global interest in this fly species (e.g., Rezende et al. 2018, Boiocchi et al. 2019, Pour et al. 2019). Here, we highlight some priority research areas with hope of renewing research efforts toward this pest insect.

One striking feature of *F. canicularis* is that its pest status throughout the United States is not universal. A well-documented pest in CA and PA, *F. canicularis* is present but rarely a pest in FL, NY, NC, GA, and TX (personal communications with J. Hogsette, D. Rutz, W. Watson, J. Hunter, and M. Merchant). Similarities in animal production type, animal husbandry, or environmental conditions are not obvious factors linking these two hot spots, and further studies may help to shed light on the inconsistent pest status of this fly.

Although the diel activity of adult *F. canicularis* has been studied (Lewallen 1954, Anderson and Poorbaugh 1964), there is little information on how environmental factors affect this activity other than the reported minimum temperature threshold for flight activity (Nieschulz 1935). Furthermore, as male swarming activity is the principal cause of public nuisance related to this fly species (Meyer 1993), environmental factors and site characteristics that impact swarm formation should be evaluated for human interventions, such as the use of fans or application of spatial repellents, to reduce fly nuisance in target areas. Diel activity patterns also presumably drive periods of fly dispersal from development sites and into surrounding communities. These drivers and related behaviors of dispersal are also unknown and require study. The last major *F. canicularis* behavioral studies examined the chasing and mating behavior of male flies and were conducted over 40 yr ago (Land and Collett 1974, Zeil 1986).

To date, no significant control methods have been developed to prevent or disrupt the dispersal of *F. canicularis* from development sites, primarily due to a glaring lack of knowledge regarding the behavior of this fly. Studies are needed to understand the distance, flight dynamics, behavior, and visual or chemical cues used during dispersal from developmental sites. Understanding these dispersal behaviors may allow for the development of management tactics that exploit the flies' natural tendencies.

There is currently no standard method for monitoring adult *F. canicularis* activity. Research on filth fly monitoring has been focused primarily on the house fly (Gerry 2020, Geden et al. 2020). Currently, sticky fly ribbons are the most common method used for surveillance of this species (e.g., Axtell 1970), but additional work is needed to identify the most effective placement locations and to determine how many fly ribbons are needed to provide appropriate sampling precision (Karandinos 1976). Sampling precision is particularly important when fly activity is nearing a level at which negative impacts are expected and where management of flies is most critical (an 'action threshold'; Lysyk and Moon 1994). Unfortunately, there is no consensus in the scientific community as to what an appropriate action threshold should be for *F. canicularis*. One challenge to determining an action threshold is that public nuisance is a difficult impact to measure, as the level of nuisance is likely to vary by both community and individual

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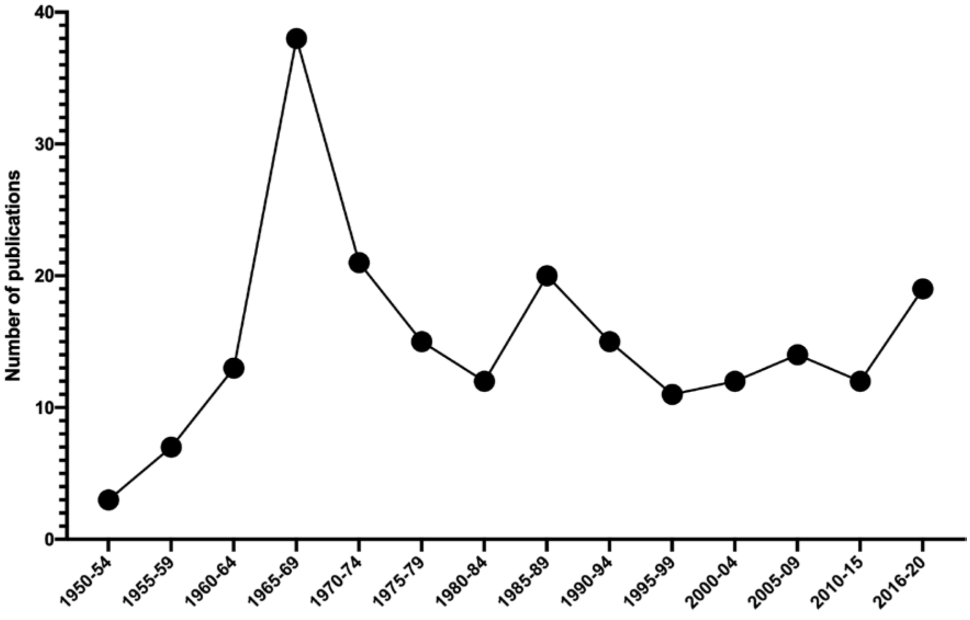

Fig. 5. Publications where '*Fannia canicularis*' was a study organism (search via Google Scholar, July 2020). Publications peaked in the 1960s followed by sharp decline. Recent published work (2016–2020) has included surveys of synanthropic flies outside of the United States, particularly those associated with forensic studies. By comparison, a search for '*Musca domestica*' for just the last time period (2016–2020) yielded over 200 results.

tolerances to the presence of flies (see Gerry 2020). Additionally, fly monitoring is currently recommended as a tool for the prevention of *Salmonella* Enteritidis in egg production (FDA 2011). Nevertheless, additional effort toward understanding *F. canicularis* nuisance and developing an accepted standard value for an action threshold is needed.

The use of granular fly baits for adult fly control is well documented for *M. domestica*. This is an inexpensive, easy to use, and target-specific management tactic (Darbro and Mullens 2004). Although fly baits do kill adult *F. canicularis* (Murillo et al. 2015), they do not attract *F. canicularis* in the field. The addition of attractants to bait stations, such as ethanol in the form of fermented carbohydrates (e.g., Hwang et al. 1978), is necessary to attract adult *F. canicularis*. Homemade bait stations using molasses and yeast to produce ethanol have been shown to attract and kill *F. canicularis* in the field (Fig. 6), but this concept requires further testing and refinement in the field before it can be widely adopted.

Additionally, the poultry industry, especially egg-layers, is undergoing shifts in housing styles and husbandry practices in the United States. Animal welfare concerns are driving housing changes in egg production away from cage systems and toward cage-free or even free-range systems (Lay et al. 2011, Murillo and Mullens 2016) thereby reducing accumulated feces that remain undisturbed by bird activity. The transition to cage-free housing styles for egg production may help to decrease fly problems as birds will disturb manure through pecking and scavenging behaviors; however, some cage-free styles such as aviaries still have areas that allow for the accumulation of undisturbed manure. Also, immature *F. canicularis* are cryptic and may not be readily consumed by poultry if they are not seen by birds (A. Murillo, personal observation). Understanding how poultry management might influence larval development sites will help to inform future control methods as well as possible cultural control tactics.

Finally, *F. canicularis* has been identified as a priority for genome sequencing and annotation. Once the *F. canicularis* genome has been sequenced, countless molecular tools currently utilized to elucidate basic and applied biological questions in other dipteran species may be utilized in addressing many of the knowledge gaps described above.

Fig. 6. Bucket traps for flies. Buckets are filled with yeast, molasses, and water, and then wrapped in screen and painted with an insecticidal fly bait. *Fannia canicularis* adults are attracted to the fermenting material, land on the treated screen where they contact the bait, and then fall into the bottom and die (photograph by Beth Futrick).

Acknowledgments

We thank the members of the S1076 regional Hatch project: *Fly management in animal agriculture systems and impacts on animal health and food safety* for manuscript discussion and advice. Follow-up edits to the manuscript provided by Luisa Domingues, Chris Geden, Jerry Hogsette, Phil Kaufman, Roger Moon, Kateryn Rochon, Jeff Scott, Dave Taylor, and Rebecca Trout Fryxell. We greatly appreciate the financial support from a Penn State Extension grant to hold a workshop in Orlando, Florida, for discussion of this manuscript and others in the series. This multi-state Extension work was supported by USDA National Institute of Food and Agriculture (USDA-NIFA) Extension Smith Lever funding under Project #PEN04540 and Accession #1000356, as well as by funds distributed by USDA-NIFA to State Agricultural Experiment Stations under the Hatch Act of 1887.

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