Studies on Oxyspirura mansoni, the Tropical Eyeworm of Poultry. IV. Methods for Control[1]

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This paper, the fourth of a series, presents the results of a study made in Hawaii on the control of *Oxyspirura mansoni*, the tropical eyeworm of poultry, and of its intermediate host, the burrowing cockroach, *Pycnoscelus surinamensis* (Linn.). Previous papers in the series report on the biology of the host roach (Schwabe, 1949), the life history of the parasite (in press), and a preliminary study of its pathogenicity in domestic fowl (in press).

The life cycle of the eyeworm is such that it affords at least three vulnerable points for the institution of control measures: (1) the adult parasites may be removed from the eyes of the host by mechanical or chemical means, (2) the eyes may be rendered unsuitable for habitation by the parasites, or (3) control measures may be undertaken against the intermediate host. The feasibility of each of these methods was considered.

THERAPEUTIC TREATMENT

The adult parasites may be removed mechanically from beneath the nictitating membranes with a pair of dull tweezers. This operation is facilitated by anesthetizing the eye with either a 2 per cent cocaine or 5 per cent butyn solution. This at best is a slow, tedious process, however, which requires the services of at least two persons.

Wilcox and McClelland (1913) and Sanders (1928) suggested the following chemical treatment: Anesthetize the eye with butyn or cocaine, instill several drops of 5 per cent creolin solution, irrigate immediately with water to wash out the creolin and dead worms.

Fielding (1926) advices instillation of a few drops of turpentine, weak Condy's fluid ($KMn0_4$), or kerosene, which is allowed to remain in the eye one half hour, and then irrigation with lukewarm water or boric acid.

Although the above treatments are effective, they involve a considerable amount of labor per bird. Since the treated birds are susceptible to immediate reinfection, under most conditions of practical poultry husbandry these therapeutic measures would be economically unfeasible.

[1] Grateful acknowledgment is made to Yoshinori Tanada, Junior Entomologist, Hawaii Agricultural Experiment Station, for supplying the insecticides utilized, Dr. M. M. Rosenberg, Assistant Poultry Husbandman, H.A.E.S., for the experimental animals, and Harry Miyata, local poultryman, for his cooperation in this project.

PROPHYLACTIC TREATMENT

The life cycle of the eyeworm is such that, as far as is known, none of the common anthelmintics could be effectively utilized either prophylactically or therapeutically against the parasite.

Several years ago Hutson (unpublished manuscript) suggested surgical prophylaxis. He found that when the nictitating membranes were removed from uninfected chickens the conditions in the eyes were rendered unsuitable for subsequent habitation by the parasites. He also observed that when the membranes were removed from infected birds, the parasites were not present in the eyes after twenty-four hours.

Inasmuch as Fielding (1927) experimentally infected guinea pigs with the eyeworm, and I was successful in doing the same with rats (in press), neither of which possesses a nictitating membrane, I decided to undertake a similar experiment in an effort to corroborate Hutson's report.

I. Both eyes of a five-week old uninfected chicken were anesthetized with several drops of a 5 per cent butyn solution. The nictitating membranes were secured with hemostats and were removed with a pair of sharp scissors. Only slight hemorrhage developed and no further treatment was necessary. The bird experienced little discomfort during or after the operation, and in several days the eyes were completely healed. One week later approximately twenty infective larvae were introduced into the mouth of the bird by pipette. Examination of the eyes at necropsy, 48 hours after introduction of the larvae, revealed that neither nictitating membrane was completely removed and that larvae inhabited both eyes.

II. The right eyes of two uninfected chicks were nictitectomized, this time special care being taken to see that the membranes were completely removed. The left eyes were to serve as controls. The chicks were exposed to infection by feeding them each approximately ten infected roaches one week after the operation. At necropsy a week later the left eyes of both birds were found to be harboring many larvae, while a careful search of the right eyes revealed no larvae. Similar results were obtained upon repetition of this experiment.

This apparently verifies Hutson's observations. The following explanation is ventured. There was evidence of adhesions of the tissues of the right conjunctival sacs of both birds as a result of the nictitectomy. A possible mechanical block to the larvae may thereby have resulted from the operation. Increased lacrimation observed in the operated eyes after removal of the membranes supports this hypothesis. Although the chickens experienced no apparent ill effects following the nictitectomy this aspect should be investigated further before surgical prophylaxis is recommended.

CONTROL OF THE INSECT VECTOR

Effective control of *Pycnoscelus surinamensis,* the intermediate host, at present appears to be the most practical and economical approach to eyeworm prevention. Although several control measures for the Surinam roach have been advocated previously, the literature concerning its life history and biology is extremely scant. As a preliminary to this work a brief study of the life history of the roach was undertaken (Schwabe, 1949). From this study it was evident that mechanical, chemical and biological control measures might be instituted against the roach. Each of these was considered.

MECHANICAL CONTROL OF THE ROACH

BATTERY HOUSING OF POULTRY:

In Hawaii, poultry is battery-raised on wire above the ground. This practice is made necessary by the shortage of available farming land and its high cost. Housing of chickens in this manner when properly done should serve as an effective mechanical barrier to the Surinam roach. I do not believe that the numbers of roaches which would reach the chickens by flying would present a problem. Each of the legs of the batteries should be set in a can of fuel oil, thereby preventing the roaches' access to the battery cages. This simple inexpensive method should prove a quite satisfactory control under most conditions.

SANITATION:

Through cleanliness and strict sanitation the roach population on the farm may be considerably reduced. The removal of accumulated manure, trash, and loose top soil from beneath the batteries destroys the habitat of the roaches. The effectiveness of this practice may be illustrated in the case of the University of Hawaii poultry farm. I made collections of roaches there in the fall of 1948. At that time manure was allowed to accumulate beneath the batteries. The roaches were numerous; a single trowel-full of soil would yield an average of thirty or more. Since that time the practice of frequent removal of the loose top soil and manure has been followed. In May, 1949, another collection of roaches was attempted there. At this time a fifteen minute search of the whole farm revealed only two adult roaches and one nymph.

The above practices of proper housing and strict sanitation are strongly recommended for eyeworm prevention.

BIOLOGICAL CONTROL OF THE ROACH

In Hawaii the natural enemies of the Surinam roach obviously fail to serve as an effective biological control.

PREDATORS:

The roach is eaten by several of the local birds, notably doves (*Streptopelia chinensis*), sparrows (*Passer domesticus*), and mynahs (*Acridotheres tristis*), all of which act as reservoir hosts of the eyeworm. In addition Illingworth (1941) found that *P. surinamensis* comprised from 40 per cent to 90 per cent of the diet of the introduced toad, *Bufo marinus*. These predators no doubt play an important yet indecisive role in reducing the numbers of the roach.

Ants are probably the chief insect enemy of the roach in Hawaii. On several occasions swarms of ants have been uncovered in the soil, which were attacking living roaches.

PARASITES:

In 1941 Williams successfully introduced into Hawaii from New Caledonia the bright green ampulicid wasp, *Ampulex compressa* F. This wasp is known to seek out and parasitize roaches of the genus *Periplaneta*. It has become well established here since its introduction.

Although I felt that the burrowing habits of the Surinam roach were such that it was not likely prey for the wasp, it seemed desirable to determine whether *Ampulex* would parasitize the roach under any circumstances. Through the courtesy of E. W. French, Graduate Assistant in Entomology, University of Hawaii, ten adult wasps were obtained. These were introduced into a large battery jar containing approximately twenty adult and nymphal forms of *P. surinamensis*. According to Williams (1942), *Ampulex* attacks its normal host immediately upon sight. The wasps were closely observed for several days, during which time they made no effort to parasitize the roaches. By the fourth day the wasps were all dead. The roaches were kept under observation for three weeks and then dissected. None showed any evidence of parasitism by the wasp.

Another group of four adult wasps was introduced into a large battery jar containing two adult *Periplaneta americana* and three adult *Pycnoscelus surinamensis*. Two of the wasps immediately attacked the American roaches, and although they were kept under observation for three days, they made no efforts to parasitize the Surinam roaches. These observations would indicate that this wasp does not parasitize *P. surinamensis*.

Hoffman (1927) reported 40 per cent parasitism of the Surinam roach by *Sarcophaga sternodontis* in the West Indies. At my suggestion C. E. Pemberton, Entomologist, Experiment Station, Hawaiian Sugar Planters' Association, a member of the Territorial Board of Agriculture and Forestry, secured authorization for the importation of this fly into Hawaii. R. H. Van Zwaluwenburg, Associate Entomologist, Experiment Station, Hawaiian Sugar Planters' Association, requested a shipment of the parasites from the University of Puerto Rico Agricultural Experiment Station. Entomologists at that station, in replying to the request, stated that Hoffman had been incorrect in his earlier observations. They reported that *Sarcophaga sternodontis* had been recovered only from dead roaches and many other dead insects, and was therefore considered saprozoic rather than parasitic. It has never been reared from a living insect as far as they were aware.

OTHER PARASITES AND COMMENSALS:

Below is a list of parasites and commensals which have been observed in or on the roaches dissected in the field and laboratory during the course of this study.

Protozoa—Numerous cephaline gregarines (not identified), occurring singly and in syzygy in the digestive tract. (These were found even in the new-born nymphs).

A small flagellate and a large ciliate (not identified) were common in the digestive tract and Malpighian tubules.

Helminths—Other than the eyeworm, no helminth parasites were found.

Insecta—No evidence of parasitism by insects was noted.

Acarina—The mite shown in figures 2 and 3 commonly attacked the roach both in the laboratory and its natural habitat. A number of laboratory-housed roaches apparently died from infestation with these mites.

No other reports of parasites of the Surinam roach have been discovered in the literature during the course of this study. Until such reports are forthcoming, prospects for the biological control of *P. surinamensis* are not encouraging.

CHEMICAL CONTROL OF THE ROACH

Alicata (1938) reported the effectiveness of butyric fermentation baits in trapping the roaches, but methods such as this would prove impractical under normal farming conditions.

Carbon bisulphide and Diesel oil have also been employed with varying degrees of effectiveness.

In 1947-48 Kartman, Tanada, Holdaway, and Alicata (1949) undertook an investigation on the chemical control of arthropod vectors of poultry parasites. Of the insecticides compared in these preliminary tests, they found parathion to be most effective against the roach, achieving a kill of 95 per cent in 24 hours, under the conditions of the experiment. Chlordane with a kill of 70 per cent, D.D.T. in kerosene with a kill of 75 per cent, and benzene hexachloride, gamma isomer, with a kill of 65 per cent, also showed promise.

Although extensive field trials of these insecticides are indicated, preliminary trials show that weekly dusting with 1 per cent benzene hexachloride, or better, spraying with 1 per cent chlordane and/or 1 per cent D.D.T. in kerosene, would materially reduce the roach population on the farm. If frequent removal of the manure were also undertaken, virtual eradication of the roach population should be the result. This treatment would probably control most of the other manure-inhabiting insects as well. This author cannot recommend the use of parathion by the inexperienced farmer or technician because of its extreme toxicity to warm-blooded animals.

SURVIVAL OF EYEWORM LARVAE

The Poultry Husbandry and Entomology Departments of the Hawaii Agricultural Experiment Station recently undertook a joint project to determine whether floor-brooding of chicks, a heretofore impractical method in Hawaii, would be possible through effective chemical control of the Surinam roach (unpublished work). The prospect that chicks might still have access to dead roaches even under such conditions of chemical control suggested the following problem.

It seemed desirable to ascertain whether or not the infective eyeworm larvae were capable of surviving any appreciable length of time in roaches which had died or had been killed by various means. Sanders (1928) killed several roaches (he did not state how), placed them in a moist chamber, and found that after 48 hours the roaches still contained active third-stage larvae. I decided to repeat Sanders' experiment as a starting point. Several adult roaches were decapitated and placed in a vial containing moist cotton. At the end of 48 hours the roaches were torn apart and placed in a water bath heated to a temperature of approximately 37° C. Numerous active infective larvae were shed. Since such favorable conditions of humidity are not apt to be encountered under natural circumstances, I modified the foregoing experiment as follows: several decapitated adult roaches were placed in an open container in the laboratory where they would be subject to the natural conditions of drying and decomposition. Roaches were removed at the following intervals of time as shown in Table 1 and checked for viable infective larvae.

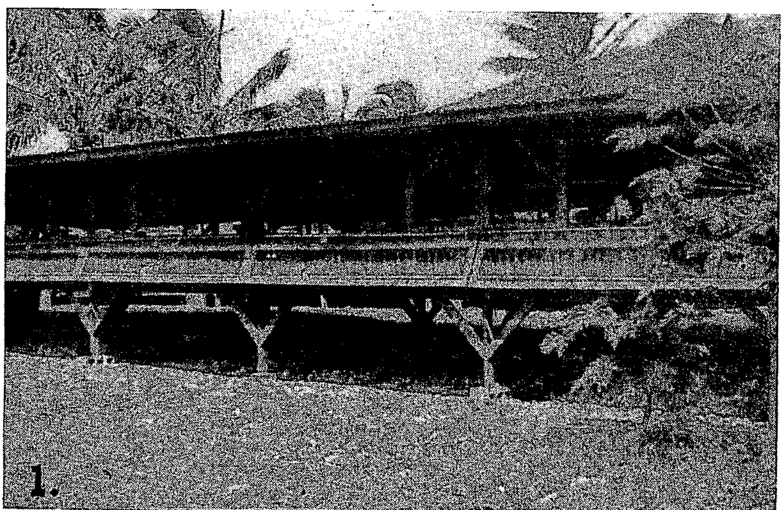
Table 1.—Survival of Infective Larvae in Roaches Killed by Decapitation

Time	Results	Time	Results
15 min.	Viable larvae shed	24 hours	Viable larvae shed
30 min.	Viable larvae shed	48 hours	Viable larvae shed
1 hour	Viable larvae shed	72 hours	Viable larvae shed
2 hours	Viable larvae shed	96 hours	Viable larvae shed
5 hours	Viable larvae shed	108 hours	Larvae dead

Having shown that third-stage larvae were capable of surviving in the body of a dead roach over an appreciable length of time under normal meteorological conditions, it seemed advisable to determine whether the larvae survived equally well in roaches killed with a variety of the common commercial insecticides.

Roaches were killed with chlordane, benzene hexachloride, sodium fluoride, parathion, and lethane by permitting them to walk over a glass surface treated with the material being tested (Schwabe, 1950). They were then placed in open containers exposed to normal atmospheric conditions and examined for viable larvae at periodic intervals. The results of these tests are shown in Table 2.

In the control roaches, death of the larvae apparently resulted from insufficient moisture and/or increased osmotic concentration. In all instances death of the larvae was hastened through exposure to the insecticides. The significant information obtained however was the fact that roaches killed with a variety of commercial insecticides serve as effective vectors for Manson's eyeworm as long as 72 hours after death.



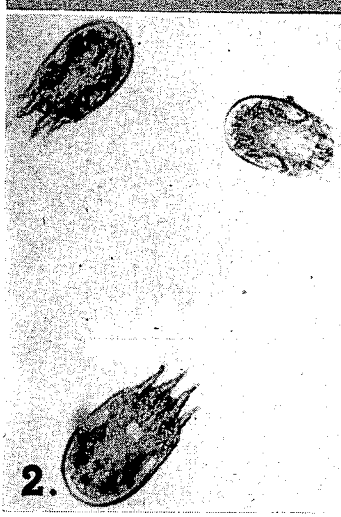


Figure 1.—The type of poultry housing and sanitary surroundings which are strongly recommended for the effective control of eyeworm infestations in domestic fowl. The legs of the batteries should preferably stand in cans of fuel oil.

Figure 2.—An unidentified mite commonly found infesting the Surinam roach, both in the laboratory and in its natural habitat.

Figure 3.—An adult *Pycnoscelus surinamensis* infested with the mites shown in Figure 2.

Table 2.—Viability of Infective Larvae—Survival when Subjected to Commercial Insecticides.

6/14 to 6/20 1949	Chlordane 2% Petroleum Solvent	Chlordane Tech. 43% Pet. sol. 47% Inert 10%	Hexachloro-cyclohexane 50% powder Inert 50%	NaF 100% dust	Parathion 15% Wettable Powder	Lethane 10% powder Inert 90%	Control Killed by Crushing the Head
12 hours.................	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed
24 hours.................	Viable larvae shed	Larvae dead	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed
36 hours.................	Larvae dead	Larvae dead	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed
48 hours.................	Larvae dead	Larvae dead	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed	Viable larvae shed
60 hours.................	Larvae dead	Larvae dead	Viable larvae shed	Larvae dead	Larvae dead	Viable larvae shed	Viable larvae shed
72 hours.................	Larvae dead	Larvae dead	Larvae dead	Larvae dead	Larvae dead	Viable larvae shed	Viable larvae shed
84 hours.................	Larvae dead	Larvae dead	Larvae dead	Larvae dead	Larvae dead	Larvae dead	Viable larvae shed

SUMMARY

The various therapeutic treatments which have been proposed for eyeworm infections in poultry are discussed and evaluated. For practical control they are not recommended since the chickens are susceptible to immediate reinfection following the treatment.

A method of surgical prophylaxis proposed by Hutson is discussed in light of experiments performed by the author, but this method is not recommended at present.

Consideration is given to various methods for the mechanical and chemical control of *Pycnoscelus surinamensis,* the intermediate host of the eyeworm. Proper housing of chickens, strict sanitation, and use of certain of the recently tested insecticides is strongly advocated by the author as the most practical and effective means for the control of the eyeworm.

The present outlook on the possibilities for biological control of the roach in Hawaii is also discussed, and data on the survival of eyeworm larvae in roaches killed by mechanical and chemical means are presented.

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