ratory at Lunds University, Sweden. Upon examination only 2 larvae from the infected site were found to be free of parasites. These were reared to adults and produced one male and one female of *Ae. hexodontus*. Ten larvae which were dissected had multiple infections, 2–5 worms occurred in each. All of the remaining larvae died upon emergence of the worms.

As the worms emerged they were transferred to jars containing clean, damp, baltic sand. Some of the worms which emerged quickly became covered with a fungus mycelium; an estimated 20% of the worms died from these infections. The *Ae. hexodontus* larvae were kept at 20–25°C until all worms had emerged. The baltic sand containing the mermithid worms was then transferred to a cold room and stored at 8–10° C. On July 25, 1978, in preparation of the senior author's return to the United States, the majority of living nematodes remaining in the baltic sand were heat killed in water and fixed in F.A. 4:1 (Seinhorst 1962).

The preserved nematodes and baltic sand were examined at R.U.V.P. in October 1978. The nematodes were processed (Baker 1953) and mounted in glycerine for identification. Unfortunately, no adult nematodes were found among the preserved specimens; 25% of those fixed had just started to molt to the adult stage. Only 4% of those fixed were immature females, the rest were immature males. This predominance of males is commonly observed when multiple infections occur in the larval stage of mosquitoes. There was evidence of fungal infections on the cuticles of 31% of the nematodes fixed. This may have been partially responsible for the failure of maturation of any worms remaining in the baltic sand, examination of which for additional worms or eggs proved negative.

As complete identification of mermithid nematodes is only feasible when adult specimens are available, it was not possible to accurately identify the specimens. However, all possessed a rounded tail and a long, sharply pointed, conic tail appendage similar to that found in species of the genus *Gastromermis*.

It is hoped that additional collections can be made from this Norwegian locality and a complete description will be published when the adult nematodes are obtained. Not only is there a strong likelihood that it may be a mermithid new to science, but its establishment in an austere arctic-type tundra environment merits its study as a potential biological control agent against the hordes of *Aedes* mosquitoes which inhabit the arctic regions in areas where man has settled.

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COLONIZATION OF *CULICOIDES WISCONSINENSIS* JONES (DIPTERA: CERATOPOGONIDAE)

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Culicoides wisconsinensis was described by Jones (1956), who collected immatures from a peat-muck substrate in Wisconsin. Jamnback (1965) recovered larvae from soft mud in a brackish marsh and a freshwater lagoon in New York State. Rowley (1967) and Wirth (personal communication) have reared this species from the margins of saline and alkaline lakes in Washington State and the Midwest, respectively. Very little else is known of its biology.

On 5 May 1980, *Culicoides* pupae were collected from a cattail bog polluted with milk-center effluent on Hutchinson Farm, St. Lawrence Co., NY. Reared females held without access to blood oviposited on wet filter paper, and the species was subsequently colonized. Specimens were held at 22°C at a 14:10 (L:D) photoperiod to gather life history data. The colony is currently in the 7th generation.

Follicle maturation in females dissected within 30 min. of eclosion was at late stage III. The pre-oviposition period was 2–3 days, during which time the females mated on contact with males in 237 ml screened holding containers or in the confines of an aspirator

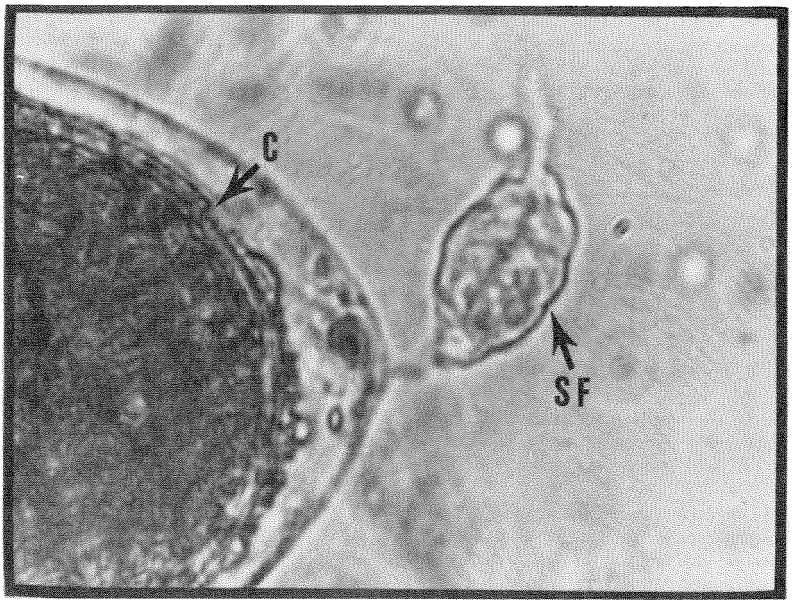

Fig. 1. Follicles of *Culicoides wisconsinensis* (× 1,000). Note undifferentiated secondary follicle (SF) above stage V primary follicle (C = chorion).

tube. Vitellogenesis took place with or without sugar, and eggs were laid on the surface of 1% agar used as a larval substrate (Kettle et al. 1975, Koch and Axtell 1978).

The egg stage lasted 3 days; eyespots were often visible through the chorion on day 2. Larvae were fed at 3–5 day intervals with an impure nematode slurry of *Panagrellus* sp., which were reared on oatmeal and yeast. All larval stages fed on the nematodes. Oogenesis produced 43–114 eggs/female ($\bar{x}$ = 76.9 ± 25.9, n = 15). Time required for development from egg to pupa ranged from 19–109 days ($\bar{x}$ = 49.3 ± 15.4, n = 385). The pupal period required 3–4 days.

To date neither nulliparous nor parous females have shown any interest in taking blood from a human forearm or, on one occasion, from a suckling mouse. Secondary follicle development remains very rudimentary in this species (Figure 1), and females usually die within a few days of oviposition, even when supplied with a sugar source. Jamnback (1965) noted the short proboscis and poor dentition of *C. wisconsinensis* and postulated that the species was not hematophagous. Our observations confirm autogenous oogenesis in specimens from northern New York and suggest that females may complete only a single gonotrophic cycle.

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AN AIRFLOW APPARATUS FOR SELECTING FEMALE MOSQUITOES FOR USE IN REPELLENT AND ATTRACTION STUDIES[1,2]

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An important consideration in our studies of mosquito repellents and attractants has been to minimize handling of the test insects when selecting and transferring them to the olfactometer for bioassays. Methods commonly used for sexing, counting, and transferring mosquitoes and other test insects for experimental purposes include the use of carbon dioxide, cold rooms, chilling tables, anesthetics, and portable electric aspirators. Some of the methods and their advantages and disadvantages have been discussed by Harris et al. (1965), Gjullin and Bevill (1972), and Magnarelli (1975). These techniques usually entail excessive handling, complete incapacitation with CO_2, exposure to chemicals such as chloroform and ether, or subjection of the insect to extreme temperature changes. When mosquitoes that have been handled by these methods are then used in the bioassay of attractants and repellents, one could assume that behavior patterns might be altered and that mortality rates may increase over normal rates (Harris et al. 1965). To avoid these problems when counting and transferring female mosquitoes to the olfactometer, a mosquito-attracting airflow apparatus that encloses a mosquito stock cage was designed that incorporates the operation principles of the olfactometer described by Gouck and Schreck (1965) and Schreck et al. (1967).

METHODS AND MATERIALS

The airflow apparatus (Fig. 1A) is 51.5 cm long, 41.5 cm high, and 40.9 cm wide. Its size is determined by the size of the stock cage (37.5 × 38.5 × 46.4 cm), as shown in Figure 1B, which is placed inside. These dimensions may be adjusted to the size of the stock cage used. The airflow apparatus is constructed from 0.6-cm thick Plexiglas® panels joined with acrylic cement and pan-head screws. After airflow apparatus assembly, a 5.1-cm wide section is cut off the front end to make a door, which is attached with a piano hinge and 2 suitcase latches to secure it shut (Figure 1C). This permits access for insertion and removal of the mosquito stock cage. A 13-cm diameter porthole (Figure 1D) is cut in the center of the

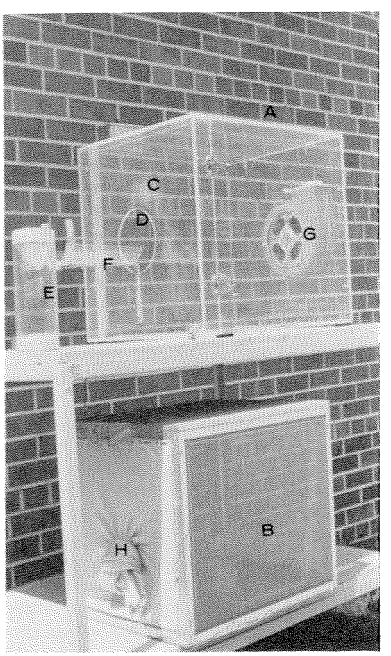

Fig. 1. Airflow apparatus (A), the stock cage (B), and the trap (E).

[1] This paper reports the results of research only. Mention of a commercial or proprietary product in this paper does not constitute an endorsement of this product by the USDA.

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