water must be carefully strained to prevent the introduction of additional larvae. After a sufficient length of time to allow the larvae to resurface (2-3 minutes for summer broods of *Ae. sollicitans*), the first dip is taken and the number of larvae dipped is recorded on a 3 × 5 in. card or other suitable paper. A total of 3 (or preferably 5) dips per sample site, with the number caught in each dip recorded, is sufficient to provide adequate data for an accurate population estimate. The field data are then used to calculate a population estimate for the sample site by the removal sampling method described above. The length of time required per sample site in the field is 5-10 minutes depending upon larval density, availability of larval escape habitat, etc. An additional 1-2 minutes is required to calculate the population for each sample site by using the method of Hayne (1949), or 3-5 minutes to calculate the regression line by the linear regression equation.

One question which frequently confronts the field investigator conducting a sampling program is the number of samples to be taken. The number of sample sites visited will depend upon area size, area of water surface providing larval habitat, time available for sampling, and the degree of precision required (as the sample size increases, results will more truly reflect the actual population, thereby increasing accuracy). When the desired number of samples has been taken and the individual population estimates for all sample sites derived, an average value for the population estimate should be calculated and expressed as the number of larvae per surface area enclosed by the area sampler. It is then desirable to calculate a coefficient of variation (C.V. = standard error × 100/ population estimate), confidence intervals, standard error, etc., as described in statistical texts such as Steel and Torrie (1960). If high values for the coefficient of variation or standard error are found, it is indicative that the sample mean is not accurately reflecting the true population mean, and the sample size should therefore be increased.

The average population estimate can be expressed in various ways. The simplest approach is to express the data in terms of density of larvae per area enclosed by the area sampler, or to multiply by the appropriate conversion factor to get larval density per square foot, square meter, etc. It is possible to derive an estimate of the larval population on a given area of marsh, woodland pool, etc. To do this, it is necessary to estimate or measure the surface area of water which provides breeding habitat and multiply by the average density figure.

To date, this technique has been used exclusively on salt marshes, where the substrate is mud and peat which facilitates rapid and firm seating of the area sampler. In a woodland pool or swamp habitat, seating the sampler may be more difficult due to the presence of sticks and leaf mats. If this type of situation is encountered, it will be necessary to have a sampler with a reinforced, sharpened lower edge, or one with saw-like teeth on the lower edge as suggested by Service (1976).

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OBSERVATIONS ON LARVAL *AEDES AEGYPTI* (L.) AS SCAVENGERS

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In our routine rearing of *Toxorhynchites brevipalpis* Theobald, individual larvae are placed in styrofoam bowls containing tap water. *Aedes aegypti* (L.) larvae are used as food and added in abundance, so that there are usually 10-20 in each bowl. On several occasions one of us (R.S.) observed that *Ae. aegypti* larvae were apparently feeding on dead *Tx. brevipalpis* larvae.

We performed the following experiment to determine (1) if the rate of disintegration of *Tx. brevipalpis* larvae, an indicator of both feeding and natural decomposition, is faster with or without *Ae. aegypti* larvae and, (2) to determine whether the *Ae. aegypti* larvae feed on dead *Tx. brevipalpis* larvae even in the presence of powdered larval food.

Fourth instar *Tx. brevipalpis* larvae were killed by freezing. Upon thawing each larva was exam-

ined with a dissecting microscope to check for any breaks or tears in the cuticle. If such were found, the larva was discarded. One larva was placed into each of 4 styrofoam bowls containing 150 ml of tap water. One bowl was left with only a *Tx. brevipalpis* larva while to the other 3 were added 15 3rd and 4th instar *Ae. aegypti* larvae. To 2 of the 3 bowls with *Ae. aegypti* larvae a pinch of larval food (a mixture of pablum, baker's yeast, fox breeder starter, alfalfa, and fish meal) was added each day of the experiment. The bowls were left on a laboratory bench at room temperature and observed morning, noon, and evening for 4 days. The experiment was repeated 4 times involving a total of 16 *Tx. brevipalpis* larvae.

We observed over the test period that in tap water the *Tx. brevipalpis* larvae did not disintegrate appreciably; an elongation of the cervical membrane was the most noticeable change. In the bowls with *Ae. aegypti* larvae, but no larval food, an average of 50% of the contents of each *Tx. brevipalpis* larva was gone within the first 24 hr. By the 2nd day 80% was gone, by the 3rd over 90%, and by the 4th 100% had disappeared. In the bowls with powdered larval food 100% of the contents of the *Tx. brevipalpis* larvae were also gone by day 4. However, the rate of disappearance was slower, i.e., 25% gone by day 1, 55% by day 2, and 85% by day 3. This suggests that starved *Ae. aegypti* larvae may feed more aggressively.

During the experiments, the *Ae. aegypti* larvae congregated around the *Tx. brevipalpis* larvae moving their mouth parts vigorously. The *Tx. brevipalpis* larvae were consumed in a stereotyped manner. The head was severed from the body by biting through the cervical membrane. The *Ae. aegypti* larvae then consumed the contents of the body and head capsule, entering both to do so. Fig. 1 shows the empty body cuticle of a *Tx. brevipalpis* larva and Fig. 2 shows *Ae. aegypti* larvae within the head capsules of *Tx. brevipalpis* larvae. Apparently *Ae. aegypti* larvae are unable to chew and ingest the body cuticle or head capsule.

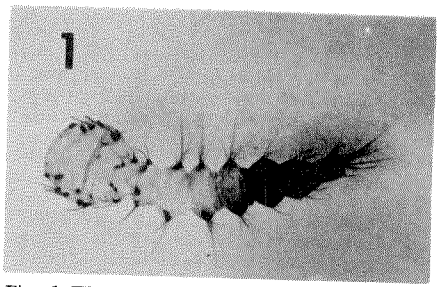

Fig. 1 Thorax and abdomen of a 4th instar *Tx. brevipalpis* larva. *Ae. aegypti* larvae have consumed the contents of the thorax and most of the abdomen leaving empty, transparent cuticle. X5

To determine if *Ae. aegypti* would consume their own kind, 12 frozen 4th instar *Ae. aegypti* larvae were placed individually into styrofoam bowls containing 150 ml of tap water and to 10 bowls, 15 3rd and 4th instar larvae were added. Dead *Ae. aegypti* larvae disintegrated much faster than dead *Tx. brevipalpis* larvae. After 36 hr the dead control *Ae. aegypti* larvae, although intact, were extremely fragile and fell apart when picked up with blunt forceps. Among the test larvae the results varied from the entire larva disappearing (50% of the cases) to a portion of the abdomen and head capsule being left. Because of size, the *Ae. aegypti* larvae did not enter the dead larvae of their own kind as occurred routinely with the *T. brevipalpis* larvae; rather they tore the larvae apart which was made possible by their rapid disintegration and hence extreme fragility.

Our observations indicate that *Ae. aegypti* larvae will feed on dead *Tx. brevipalpis* larvae and *Ae. aegypti* larvae and will ingest the contents of the former in the presence of abundant powdered larval food. These observations are an additional example that *A. aegypti* larvae are opportunistic feeders, probably capable of utilizing a wide variety of organic matter.

Our observations are concerned with *Ae. aegypti* as scavengers. However, they have been observed in certain situations to feed on live larvae of their own kind. Older larvae of both *Ae. aegypti* and *Anopheles stephensi* Liston are

Fig. 2 *Ae. aegypti* larvae within head capsules of *Tx. brevipalpis* larvae. X3

known to be cannibalistic (MacGregor 1915, Reisen and Emory 1976). With *An. stephensi* the small larvae were seen to be caught in the currents created by the action of the mouth brushes of the older larvae. Reisen and Emory (1976) also observed that older *An. stephensi* larvae would seize one another with their mouth parts, but soon separated.

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THE OCCURRENCE OF *AEDES SOLLICITANS* IN WESTERN NEW YORK

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Inland distribution records for *Aedes sollicitans* are rare. Breeding sites other than in coastal saltmarshes have been reported for this species by Fellton (1944). The collection of adult specimens of this saltmarsh mosquito in western New York led to an investigation which located a unique but not uncommon, breeding habitat for this species. A description of the mosquito breeding site, and the possible significance of this new habitat as a focus for a mosquito-borne disease outbreak are discussed.

The saltmarsh mosquito, *Ae. sollicitans,* is an important pest species and a potential vector of arboviruses. Its distribution is primarily confined to the coastal marshes of the southeastern counties of New York and Long Island. It has been collected upstate in Onondaga County by Barnes et al. (1950).

Adult specimens of *Ae. sollicitans* were collected with an aspirator on August 21, 1972 and again on August 31, 1972 at the Town of Amherst in Erie County. This constitutes a new distribution record in New York. The breeding site for this species was located following an extensive search in late 1972 and 1973. A prolific and versatile mosquito breeding site was found adjacent to a New York thruway exit ramp during the late summer of 1972. Larvae of *Culex salinarius, Cx. restuans,* and *Cx. pipiens* were identified from this site in September of 1972. In the spring and early summer of 1973, larvae of *Ae. stimulans* and *Ae. dorsalis,* and adults of *Ae. dorsalis* were collected at this site. On July 19, 1973, 3rd and 4th stage larvae of *Ae. sollicitans* were found at the site.

This mosquito breeding site was a 100-ft. ditch with a sandy bottom located at the base of a grassy embankment leading up to a New York State thruway exit ramp. The slopes of the embankment showed visual evidence of road salt which remained from winter snow dispersal methods utilized during the previous winter. The ditch was about 3 ft wide, and the water depth never exceeded 2 ft. During the dry season about two-thirds of the ditch was dry. The water in the ditch was rich in organic matter, and was continually fed by an overflow of the septic tank and its corresponding sand filter from the toll station office.

Water samples were taken from the ditch biweekly from June 21, 1973 to September 27, 1973, and analyzed chemically and biologically. The pH ranged from 7.4–8.4. The salt content of the water was found to be elevated in the early summer when the *Ae. sollicitans* larvae were found. The salt levels were lower in the fall when the *Ae. stimulans* and *Ae. vexans* larvae were present. These salt levels ranged from 670–1700 ppm for sodium, 50–2750 ppm for chlorides, and 50–165 ppm for calcium. The B.O.D. ran from 2.2 to 35 ppm. The water contained many organisms including; algae, amoeba, copepods, diatoms, and euglena. Total coliform counts ranged from "overgrown" early in the summer to 90/100 ml of sample in the fall.

Drainage problems produced by construction and maintenance procedures used along many super highways have allowed the establishment of mosquito species which have not been previously associated with an area. Normal relief drainage of surface water in many areas has been blocked, thus creating uncontrolled ponding of water. Environmental progression of this water through the addition of chemical and physical pollutants produces overgrown swamplands and bogs which are ideal breeding sites for many of the mosquito species which transmit arboviruses. Construction of subdivisions, shopping plazas, and recreational areas adjacent to these super highways is exposing a greater proportion of the population to the vectors of the arboviruses. Breeding sites which have produced only *Ae. stimulans* and *Ae. vexans* in previous years are currently yielding *Ae. sollicitans, Ae. dorsalis, Cx. pipiens* and *Cx.*