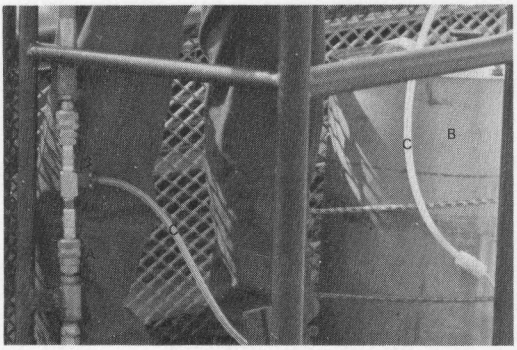

Fig. 3. Close-up of Dema injector (→) with quick-disconnects (A) and Arosurf MSF supply tank (B) with associated suction hose (C).

spray gun from the truck for repair or replacement. The airline "quick-disconnects" can be purchased from any automotive parts store.

Dema Model #202C injectors cost $31.50 each. Nozzle bushings are $2.35 each and there are 3 sizes available for this model. Airline "quick-disconnects" for the entire system cost $10.02. The truck was equipped with a 13 gal stainless steel ULV tank containing Arosurf that was mounted on the spray truck with elastic cords for easy removal and refilling. Plastic containers can also be used.

The injector system is currently being used on an experimental/semi-operational basis to dispense Arosurf MSF in water or Arosurf MSF into an Abate 4–E/water mixture. Investigations are also being conducted on the use of a two injector system that can be interchanged in and out of the system in the field. One injector would be used to apply technical insecticides such as Abate 4–E or commercial preparation of *Bacillus thuringiensis* var. *israelensis* for larviciding and the other injector would be used for application of Arosurf MSF for pupiciding. The use of computerized ULV flow controls are also being evaluated.

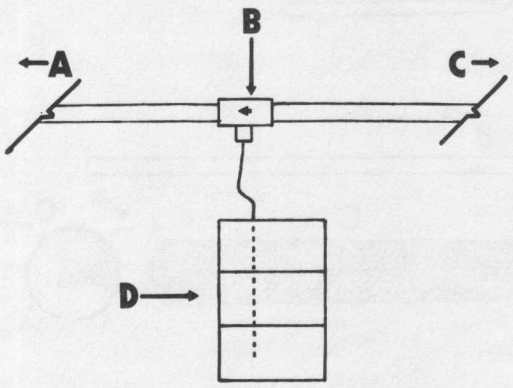

Fig. 4. Schematic representation of Dema injector placement and flow diagram. A. Pump, B. Injector with arrow indicating flow direction, C. Spray gun, D. Supply of Arosurf MSF.

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DEVICES FOR SAMPLING AND SORTING IMMATURE *COQUILLETTIDIA PERTURBANS*[1]

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Coquillettidia perturbans (Walker) are major human and livestock pests and potential vectors of arboviral diseases. Although the adults can be controlled by chemical insecticides, the larvae, which spend much of their time attached to the submerged stems and roots of emergent vegetation in highly organic habitats, generally can not (Hagman 1953, Guille 1976, H. D. Newson, personal communication). The development of alternative larval control methods has been slowed in part by a lack of knowledge of the ethology and ecology of the immatures. This is a direct result of not having a simple, standardized, cost-effective and quantative larval detection method.

A number of methods have been used to collect *Coquillettidia* immatures (McNeel 1931, Bidlingmayer 1954, Barton 1964, Morozov

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1965, Guille 1975, Gozhenko 1978, Rademacher 1979, Allan et al. 1981), but all have limited applications and are either laborious, time consuming and/or inappropriate for quantitative studies. Consequently not one has been generally adopted as a standard method for research or surveillance purposes.

This report describes methods developed in central Florida to collect mosquitoes from natural breeding sites without destroying the habitat and to separate the larvae from the detritus in the sample without destroying the larvae. These methods are appropriate for qualitative evaluations of potential breeding sites and quantitative studies on larval spatial and temporal distributions.

The collection system consists of a probe, a pump, a sieve and connecting hose. The probe (Fig. 1) is a 30″ length of 1/2″ electrical conduit with four 3/8″ holes drilled perpendicular to each other near the lower end. The lower end of the conduit is enclosed in a 2″ diam, 10″ length sleeve of 18 gauge stainless steel perforated with 5/16″ holes. A second length of conduit, which can be of variable length, is bolted to the first to serve as a handle. A garden hose connects the upper end of the probe conduit to the intake of a 12V DC self-priming pump (Jabsco Water Puppy model #6360–0001) powered by an automobile battery.

The probe is set on the bottom of the marsh or in a floating mat of vegetation. Pumping for 1 min draws up approximately 4.25 gallons of water and detritus which is directed through a 350 micron mesh nylon sieve attached to the wide end (8″ diam) of a galvanized funnel (Fig. 2). The habitat water is pumped to the narrow end of the inverted funnel. After pumping is completed the narrow end of the sieve unit is placed in the mouth of a 1 qt Mason jar and the sieved material backwashed into the jar using habitat water. The Mason jar is capped with a prelabeled lid, returned to a carrying case and covered to prevent heat buildup from the sun (see Fig. 3).

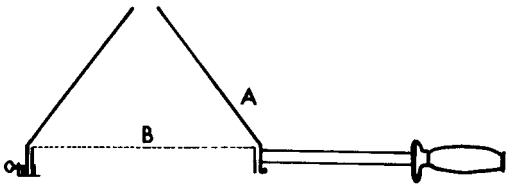

Fig. 2. Sieve unit showing the galvanized funnel with handle (A) and 350 micron mesh screen (B).

The samples are then returned to the lab for further processing. The unit (Fig. 4) used to extract the larvae from the detritus is a further miniaturization of the cylinder/cone system developed by Bidlingmayer (1954) for field collecting larvae which Lounibos and Escher (1983) modified for lab use.

The separation cylinder is a 10″ length of 3″ i.d. schedule 40 polyvinyl chloride (PVC) glued to a base of 4.5″ × 4.5″ × 0.25″ Plexiglas®. At 6.5″ above the base, three 1/8″ holes are drilled through the PVC to accommodate seating pins that protrude 1/4″ inside the cylinder. The seating pins are 3/16″ stove bolts which self-tap when screwed into the PVC. A plastic circular funnel was trimmed to fit the inside diameter of

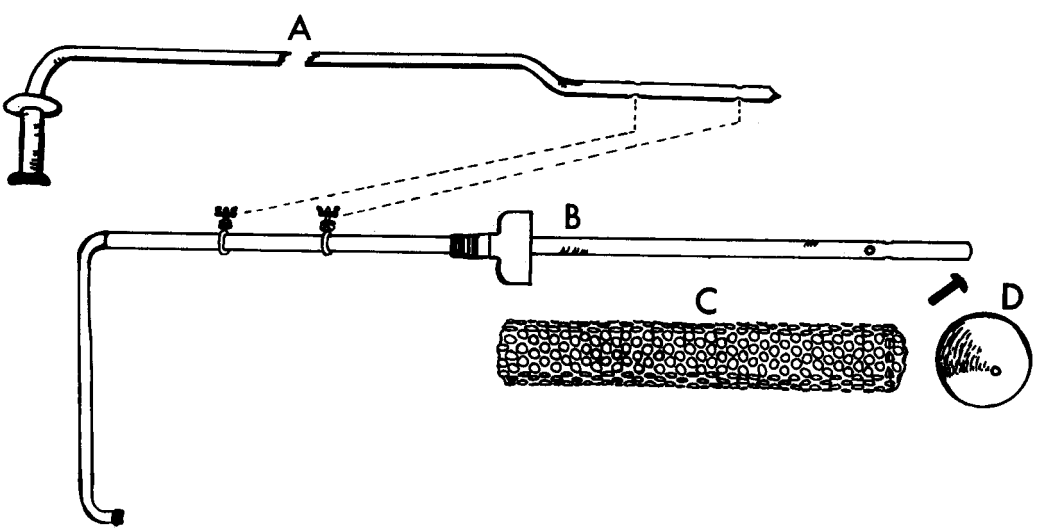

Fig. 1. Probe unit showing the handle (A), probe conduit (B), filter (C), and end cap (D).

Fig. 3. Field use of the probe system.

the PVC and the stem of the funnel was removed to create a 5/8″ diam. hole. The funnel is held in place atop the seating pins by a 9.25″ length of 3/16″ diam. plastic tubing which pressure fits between the funnel and the PVC.

The contents of a sample jar are poured into a cylinder which is then filled with water to the level of the seating pins. The funnel and tubing are then put in position before more water is added to bring the level 1/2″ above the funnel. The samples are held overnight before the ca. 200 ml of water above the funnel is poured into a pan and examined for larvae.

The probe and separation systems have been used successfully to sample a wide variety of floating and rooted vegetation in natural and man-made wetlands. Sampling is typically done from an airboat (Fig. 3), but the system is compact enough and light enough to use from a canoe or johnboat. In a day, a team of two can process 75–100 samples without difficulty. When the automobile battery is replaced with a smaller battery (Gel/Cell GC 1260 12V 6 amp) the system can be used as a backpack. The Gel/Cell will provide about 30 min of pumping.

Because of its small diameter and the ability to vary its handle length, the probe can be used to sample any aquatic habitat where mosquito larvae are likely to be found, including areas with thick roots or deep water which precludes sampling with most other systems. The electric pump standardizes the method by eliminating the variation in sampling procedures inherent when different people use a system requiring special manipulation. The standardized separation system is also more effective because it eliminates the human error and fatigue associated with the tedious and time consuming hand-sorting method.

While the pumping system was developed to sample *Coquillettidia perturbans*, we have collected the ecologically similar genus, *Mansonia*, as well. In addition, we have collected *Culex*, *Anopheles* and *Uranotaenia* in large enough numbers to suggest that these methods may also be applicable to the collection of surface breathing species which tend to dive to the bottom when disturbed. With modification, the probe may also be useful to sample species which breed in tree holes, crab holes, rock holes and

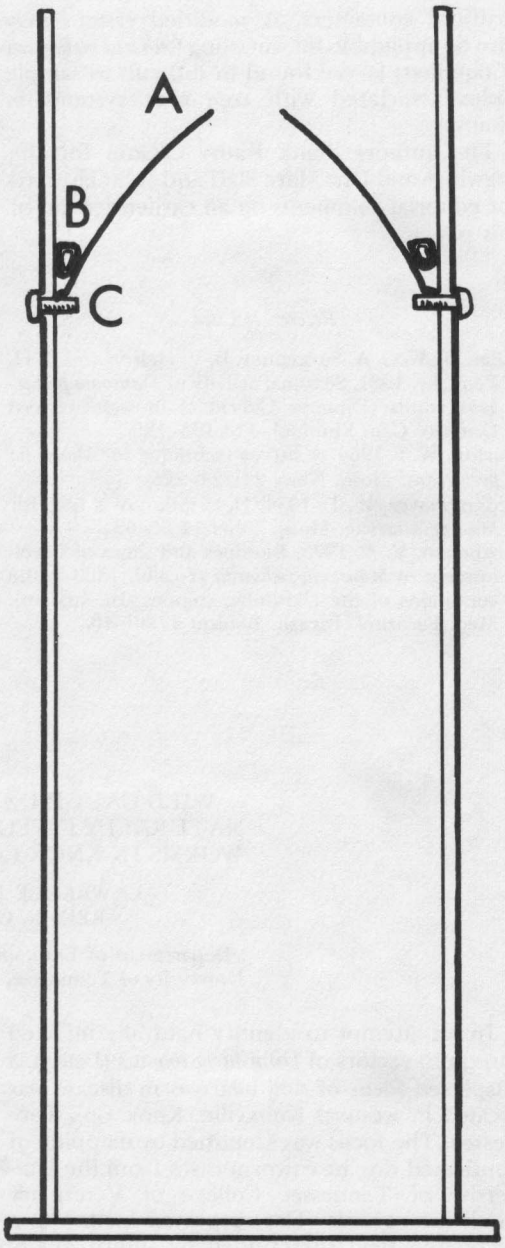

Fig. 4. Unit showing the inverted plastic funnel (A), the plastic tubing (B), and the seating pin (C).

artificial containers. A modified system may also be applicable for sampling *Culiseta melanura* (Coquillett) larvae found in difficult to sample holes associated with tree root systems in swamps.

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WILD-CAUGHT *AEDES TRIVITTATUS* NATURALLY INFECTED WITH FILARIAL WORMS IN KNOX COUNTY, TENNESSEE[1]

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In an attempt to identify naturally infected mosquito vectors of *Dirofilaria immitis* (Leidy), a suspected focus of dog heartworm disease was located in western Knoxville, Knox Co., Tennessee. The focus was identified by mapping of confirmed dog heartworm cases from the University of Tennessee College of Veterinary Medicine records. The suspected focus was a recreation field surrounded by suburban development with many free-running dogs. The primary mosquito breeding area was a wet weather pond ca 0.25 hectare adjacent to the recreational field. Mosquitoes were collected from the suspected focus using a heartworm-free dog in traps modified from Magoon (1935) and Shemanchuk (1978) from June to September 1984. Mosquitoes were identified to species, dissected in insect saline (Taylor 1960) and examined microscopically for filarial worms within 24 hr of collection.

Eight hundred and forty-four mosquitoes were dissected during this study. One of 530 *Aedes trivittatus* (Coq.) examined contained 5 L_2 in the Malpighian tubules, and 15 L_3 in the hemocoel. None of 3 *Ae. thibaulti* Dyar and Knab, 114 *Ae. triseriatus* (Say), 52 *Ae. vexans* (Meigen), 9 *Anopheles punctipennis* Theobald, 1 *An. quadrimaculatus* Say. 2 *Culex pipiens* Linn., 5 *Cx. salinarius* Coq., 1 *Psorophora cyanescens* (Coq.) and 127 *Ps. ferox* (von Humboldt) were infected.

Aedes trivittatus has been implicated as a vector of *D. immitis* in Iowa (Christensen and Andrews 1976) and Indiana (Pinger 1982). Pinger (1982) states that this species should be considered an important vector of the parasite whenever it occurs in large numbers. Live mammal trapping and examination for footprints of the sus-

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