at the places where the pins are to be inserted, since they hold firmly when the cotton has been tightly compressed. The box is lined with the wet cotton on 2, 3, or 4 sides, as desired, plus the inside of the cover optionally. The floor should be thickly lined. A small jar of water on the floor of the box keeps down evaporation. When in use, the lid is imperfectly placed over the top so that a ¼-inch slit is left at one end for air. The contents are therefore damp and dark. If desired, instead of absorbent cotton, sheets of 1-inch thick plastic foam can be pinned as a lining for holding water. Layers of soaked gauze may also be used. To wet the lining again, it is not necessary to remove it. The cages are removed and water is dropped on to the absorbent material directly from a hypodermic or rubber syringe.

Any type of small mosquito-holding cage can be inserted into the chamber, but a very convenient size is a 4-inch cubical cage, or one made from a plastic cylinder 4 inches in diameter and 3 to 4 inches high. The sides can be completely solid, if a nylon or cotton netting is stretched across the top. A sleeve may be inserted either at the side or bottom, and should be at least 8 inches long. Instead of a sleeve a piece of slitted rubber dam may be used to admit an aspirator, covering a small or large hole at the bottom or at one side. The cages may be staggered and tiered within the humidity chamber. Styrofoam or plastic foam blocks may be used to separate or tier the cages.

If it is desired to maintain the mosquitoes on sugar, a 25 percent solution of sucrose or dextrose is used. Small pieces of absorbent cotton are dipped into the solution and laid across the netting at the top. Water in which raisins have soaked for several days or been boiled, can be used in place of the sugar solution, or the boiled or soaked raisins themselves used by laying them across the top of the cage. Care must first be taken to test the raisins for insecticidal residues which may have been on the original grapes. Male and female mosquitoes are kept together in the cages, but the males usually begin to die off after 3 or 4 days. The females thrive inside this humidity chamber, which can be used in an airconditioned room.

If it is desired to infect the mosquitoes in the small holding cages by having them feed on a bird or animal having pathogenic microorganisms in the blood (virus, microfilariae, protozoa, etc.), the immobilized host is laid across the nylon netting at the top of the cage. If the donor is human, the forearm is laid upon the top netting.

The long-term maintenance of infected mosquitoes in this chamber has been successfully accomplished with *Anopheles quadrimaculatus, Anopheles stephensi, Aedes aegypti,* and *Culex pipiens pipiens,* which were infected with Friend murine leukemia virus. The first three of these species feed avidly in this type of set-up. Caged *C. p. pipiens,* however, feed sporadically, and must be well-starved in order to ensure that adequate numbers of females take up a full blood meal from the donor. If *A. aegypti* eggs are desired, strips of wet filter paper or paper towel are placed at the bottom of the cage containing the males and females. Even if this is overlooked, it was found that the females, while standing upside down feeding on the sugar-soaked cotton, laid their eggs directly on the cotton through the mesh. A small water-filled receptacle must be inserted into the cage to obtain eggs of the other species.

CONVERSION OF THE NEW JERSEY LIGHT TRAP FOR COLLECTING LIVE MOSQUITOES IN DA NANG, VIETNAM [1]

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Prior to 1969, the Entomology Department, Preventive Medicine Unit, Naval Hospital, Da Nang, Vietnam, conducted routine entomological surveillance and provided logistic support to the Navy and Marine Corps units of the area.

Surveillance of the mosquito population was conducted by operating standard New Jersey Light Traps. In June 1969, the junior author organized an *Anopheles* dissection laboratory in the Entomology Department. It then became necessary to develop a method of collecting live *Anopheles* mosquitoes for dissection studies. It was not possible due to combat conditions, lack of personnel and high incidence of malaria, to conduct night bite counts on a routine basis. Other established methods of live collecting proved impossible because of the lack of equipment. Since the New Jersey Light Traps were the collecting equipment available, it was necessary to adapt them to meet the authors' needs.

After some experimentation, the 5/16-inch wire mesh screen was removed from the opening of the cylinder of the trap. This was done because debris created by the great number of large insects striking the screen damaged or killed the smaller insects being drawn into the trap. The rotation of

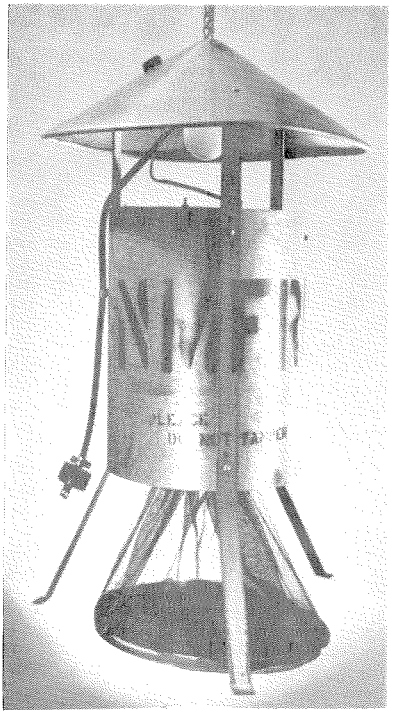
[1] The opinions or assertions contained herein are the private ones of the writers and are not to be construed as official or reflecting the views of the Navy Department or the naval service at large.

the fan blades resulted in little damage to the mosquitoes.

The kill jars were replaced with cylinders of netting similar to those found on the CDC light traps. Rubber bands held the nets in place on the traps. The collection cage was approximately 12 inches in diameter and 6 inches in height. It provided ample room for the movement of live insects. The downward pressure of the fan prevented loss of insects through the net opening. Moderate rainfall resulted in little loss of material, due to the construction of the light trap. If the nets became wet, they were dried by hanging in the sun.

More live adult mosquitoes could be returned to the laboratory if the cages were collected just after sunrise. Desiccation of the material resulted when the cages were left in the sun. Upon returning to the laboratory, the nets were placed in the freezer compartment of a refrigerator for 15 minutes. Then the contents were sorted and the mosquitoes identified. Viable *Anopheles* specimens were then dissected. In 9 months approximately 2000 *Anopheles* were dissected.

This method of live collecting produced large numbers of viable wild adult *Anopheles* for dissection. It was also inexpensive since standard light traps were used. Nets could be produced for less than $3.00. Standard military bed and head nets provided the ideal type of material for construction of the cages (See figure). Had the authors been working in less remote and primitive circumstances, other techniques would have been employed. However, the authors believe that this technique may be of future value to workers who suddenly need live specimens for some purpose when they do not have the benefit of proper collecting equipment.



TWO MOSAIC GYNANDROMORPHS OF *Culex tarsalis* COQUILLETT FROM TEXAS

F. C. HARMSTON [1]

Six gynandromorphs of *Culex tarsalis* Coquillett have been reported previously from field-collected and colonized specimens. Three field-collected, bipolar gynandromorphs, with female heads and male genitalia, are known from California, Arizona, and Texas. One field-collected bipolar specimen with male head and female genitalia is reported from Arizona. Two mosaics, displaying both male and female characters on the head, and male genitalia, are known from a field-collected specimen from Colorado and a colonized individual from the Bakersfield, California, laboratory strain.

The two gynandromorphs under consideration were collected at the Hahn Ranch, about 2½ miles northeast of Hale Center, Texas. Gynandromorph A (Fig. 1–2), was taken in a sentinel chicken shed trap, June 18, 1969. This specimen has normal female antennae; right palp, normal female; left palp, male atrophied; right fore tarsal claws, normal female; left fore tarsal claws, normal male; right middle tarsal claws, inner claw atrophied, male, outer claw, normal female; left middle tarsal claws, normal male; hind tarsal claws, normal. Genitalia normal, male.

Gynandromorph B (Figs. 3–4), was taken in a light trap, September 16, 1969. This specimen has normal antennae; right palp, atrophied male, but more fully developed than the atrophied left palp in Gynandromorph A; left palp, normal female; right fore tarsal claws, normal male;

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