

1. Dechorionate eggs in diluted Clorox and wash.
2. Fix in formol-alcohol-acetic acid, pricking the eggs with tungsten needles sharpened in melted sodium nitrite.
3. 30% alcohol, 15 minutes; 70% alcohol, 15 minutes.
4. Stain 24-48 hours in alcoholic borax carmine.\* Best results were obtained with pre-World War I German stains kindly given me by Kenneth Cooper, but carmine obtained from G. T. Gurr gives good results. Superficial staining occurs within a few hours but nuclear details are better in embryos stained for longer periods.
5. Destain with acid alcohol (a few drops of concentrated HCl in 70% alcohol) until desired contrast is obtained, usually 24-48 hours. Embryos should be a pale cherry red and will deepen in color when placed in xylol.
6. Dehydrate through absolute alcohol, clear in xylol, and mount. To prevent fragmentation of embryos, the coverslip is supported on two sides by small pieces broken from no. 1 coverslips. Sealing with fingernail polish prevents shrinkage of the mounting medium away from the sides of the coverslip.

\*Alcoholic borax carmine (after ROMEIS): Grind together finely 2-3 grams carmine with 4 grams borax. Dissolve by slowly heating in 100 ml distilled water. Cool. Add 100 ml 70% alcohol. Let stand for a week shaking frequently. Filter before use.

Spieth, Herman T.<sup>1</sup> University of California. A method for transporting adult Drosophila.<sup>2</sup>

In the course of recent investigations of the endemic Hawaiian drosophilids, some difficulties arose with the transportation via motor vehicles and/or inter-island airplanes of field captured adult Drosoph-

ila from the cool, wet rain forests to the much warmer coastal area of Honolulu. Not only are the insects sensitive to heat and desiccation, but also they are likely to become "stuck" in the food and to the walls of containers more often than is true for individuals of species from other parts of the world. The following method was devised and has proven eminently successful for the transportation of the flies not only from the field to the laboratory at the University of Hawaii in Honolulu, but also from Hawaii to the University of Texas via air freight.

Eight-dram vials are lined with water-dampened, 6.5 x 6.5 cm. pieces of chromatography paper that has approximately the same texture and thickness as that of a common desk blotter.

The following media is then prepared:

- 15 gm. Bactoagar
- 1000 ml. Water
- 50 ml. Karo syrup (dark)

The agar is added to the water and the mixture is heated sufficiently to dissolve the agar. The Karo syrup is then added and the resulting mixture is simmered for 2 to 3 minutes.

Into each paper-lined vial the hot mixture is poured to a depth of approximately 8 mm. (0.25 in.), the exact amount to be determined by the absorbency of the paper being used. The vials are tightly stoppered with non-absorbent cotton, and are then autoclaved for 15 to 20 lbs. pressure, after which the autoclave is slow-exhausted. When removed from the autoclave, all of the liquid medium should have been absorbed into the paper lining and only a very thin film should remain on the bottom of the vial.

The vials should be prepared 1 to 2 days before use but since they are sterile they do not need to be refrigerated. If they are to be kept a longer time before flies are introduced into them, they should be stored in a tight container to reduce evaporation via the cotton plug.

When the flies are introduced into the vials, they feed readily upon the surface of the media-impregnated paper and cling to the surface easily. Fecal materials are absorbed into the paper and condensation does not form upon the paper surface when the vials containing flies are placed in a cold insulated shipping container that is kept cooled by means of 2 or 3 frozen containers of "Scotch Ice," "Magic Cold," or other chilling agent. The individuals of all species tested to date have remained healthy for 4 to 7 days before it was necessary to change them to a fresh vial.

When preparing for shipment, the vials should preferably be laid on their sides in the shipping container and should be wrapped and padded with newspaper.

White blotting paper can be substituted for the chromatography paper but in our experience residual impurities in the paper, apparently localized to limited areas, cause a small percentage of the vials to become poisonous to the flies due to the release of the noxious materials as a result of the autoclaving.

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Wrathall, C. Richard and E. W. Hanly.  
University of Utah. Another plug for culture vials.

We have found in this laboratory that large rayon balls, purchased from Kendall Co., Fiber Products Division, Walpole, Massachusetts, 02081 (No. 6898, size 580) make very successful plugs for the commercially available 8 dram shell vials. Their cost is low (5.00/2000), they fit perfectly into the vial (time saving) and retain their color and resiliency after many autoclavings.

Mellett, J. S. Iona College. Plastic beakers for culturing Drosophila.

Any workers (particularly those dealing with undergraduates in genetics laboratory courses) dissatisfied with the traditional glass bottle method of culturing Drosophila

might be interested in the disposable "Multi-pour" beakers currently being marketed by Clay-Adams Inc., 141 E. 25th St., New York, N. Y., 10010. They are available in four sizes (50, 100, 250, and 400 ml), each with a tight fitting cardboard cap, on which a mass of information can be recorded. While probably all are suitable for Drosophila genetics work, I have found the 100 ml size the best for student experiments. (Fig. 1).

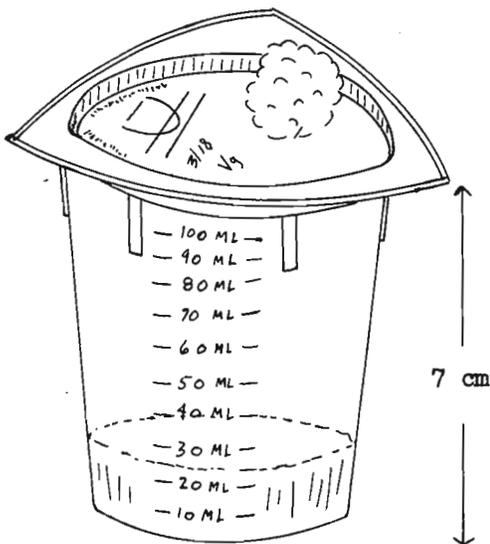


Fig. 1 Clay-Adams 100 ml "Multi-pour" beaker

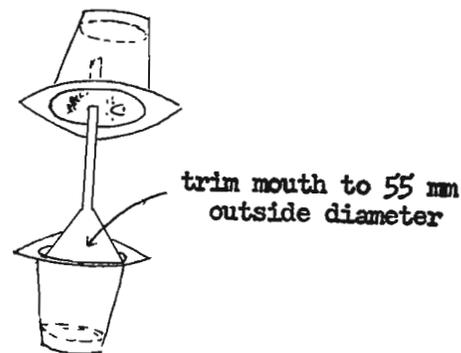


Fig. 2 Transfer method using polyethylene funnel