chloride (U.S.P.) for 20 min. Because of the aforementioned effect that alcohol has upon the plastic, tinctures must necessarily be avoided. Boiling of the device causes cloudy depolymerization and is therefore inadvisable. The ½″ height of the device permits the intact removal and subsequent replacement of the specimen cup without replenishment of the distilled water or buffer solution in which the electrodes are ordinarily maintained.

When testing minute quantities by this method it is particularly desirable that the electrodes and microbeaker be scrupulously clean and adequately rinsed. The electrodes are ideally rinsed three times and blotted with a fresh sheet of cleansing tissue. This procedure is especially recommended if the specimen should be poorly buffered. For a very critical test a contact discrepancy in readings may be avoided by employing the device with the preferred buffer solution when making the asymmetry potential corrections. It is also preferable to use a buffer solution of approximately the same pH value as the solution being tested. The plastic is comparable to soft glass as a nonconductor, possesses less porosity, and is virtually unbreakable.

A Rapid Method for Preparing DDT in the Laboratory[1]

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DDT is an abbreviation of the chemical term, dichlorodiphenyl-trichloroethane, $(C_6H_4Cl)_2CHCCl_3$. This outstanding new insecticide has become, during the last 5 years, the subject for numerous investigational studies in the entomological and chemical fields. The commercial technical-grade DDT is a mixture of several isomers and impurities (*3*). It consists essentially of 70–75% of *p,p′*-DDT, 1-trichloro-2,2-*bis* (*p*-chlorophenyl) ethane. The *p,p′*-DDT isomer is mainly responsible for the insecticidal properties of the compound. When pure, it has a melting point of 108°–109° C.

Commercially, DDT is now prepared by the Baeyer condensation method originally employed by Zeidler (*5*), who was the first to report the synthesis of this compound. The process involves the use of chloral, fuming sulfuric acid, expensive and more or less complicated apparatus (*4*). Both chloral and fuming sulfuric acid are unpleasant and hazardous to handle, especially by inexperienced workers and students. Recently a laboratory method was suggested by Darling (*2*) in which chloral hydrate is substituted for chloral but requires the addition of oleum.

Research work with DDT, as well as teaching insecticides, often requires the synthesis of small quantities of pure DDT in the laboratory. Not all laboratories are equipped with suitable apparatus to handle hazardous reagents. The writer, being confronted with such a problem, has worked out a short and simple method for preparing small quantities of DDT without the use of either chloral or fuming sulfuric acid. The entire process can be carried out within one laboratory period and has been successfully used during the last two years by the writer and graduate students taking courses in insecticides.

The method is based on the theoretical reaction of 1 gm mole of chloral hydrate and 2 gm moles of chlorobenzene (sp.gr., 1.107) in the presence of about 4–5 times their combined volume of concentrated sulfuric acid.

When only small quantities of DDT are desired, correspondingly smaller proportions of ingredients, based on 0.1 gm mole of chloral hydrate, such as 17 gm of chloral hydrate and 23 ml of chlorobenzene, are convenient to use.

The procedure is as follows:

Place 17 gm of chloral hydrate crystals and 23 ml of chlorobenzene in a glass-stoppered, 500-cc Pyrex reagent flask and keep in electric oven at 60°–70° C for about 20–30 min with occasional shaking, or until all the crystals have dissolved. Cool to room temperature and slowly add about 180 ml of concentrated H_2SO_4. Stopper and shake vigorously until precipitation starts. This operation usually requires about 1 hr. Let stand for about 15 min with frequent shaking or until precipitation is complete. The upper solid layer contains the crude DDT.

Pour the mixture into a glass jar containing about 1 gal of cold tap water and allow to stand for 15 min, or until the solids have settled. Filter through three layers of cheesecloth and wash several times with tap water. Transfer residue from cheesecloth into a wide-mouth bottle or a small glass jar, add about 50 ml of either 2% Na_2CO_3 or 4% $NaHCO_3$ solution, and shake for 5–10 min. This will neutralize the acidity.

Filter through a small, dry, Buchner funnel and wash several times with distilled water, or until the filtrate is neutral to litmus. Continue air suction for a few minutes or until no more water drains out. Transfer the residue to a small porcelain mortar, add about 100 ml of ethanol, and triturate with a pestle for 5–10 min. Filter through a dry Buchner funnel, rinsing twice with 25 ml of ethanol. Continue suction until no more alcohol drains out.

Dry the residue at 70°–75° C in an electric oven for 2 hrs, or until all the alcohol has volatilized. Cool to room temperature and weigh. An average yield of about 16 gm of practically pure DDT is obtained.

Samples from 6 batches prepared during the last two years were analyzed for *p,p′*-DDT by the chemical method developed by Cristol, Hayes, and Haller (*1*). The results have shown variations of from 94.5 to 98.35% *p,p′*-DDT, giving an average of 96.82%. The melting point ranged from 106° to 108° C.

The laboratory-prepared DDT was tested on third-instar larvae of *Aëdes aegypti* in comparison with a commercial sample of DDT (technical grade). The tests were made in beakers, each containing 200 ml of distilled water and about 50 larvae. One ml of ethanol containing various concentrations of the toxicants was dispersed in each beaker. Each concentration was run in triplicate, giving a minimum of 150 larvae per test.

[1] Paper of the Journal Series, New Jersey Agricultural Experiment Station, Rutgers University, Department of Entomology.

At the end of 48 hrs the dead and live larvae were counted. During this period some of the larvae had pupated, and the live pupae were included with the surviving larvae. The results given in Table 1 indicate that

TABLE 1

COMPARATIVE TOXICITY OF LABORATORY-PREPARED DDT AND COMMERCIAL DDT TO LARVAE OF *Aëdes aegypti*

Dilution (ppm)	Per cent larvae dead in 48 hrs			
	Laboratory		Commercial	
	Total No. larvae	Dead (%)	Total No. larvae	Dead (%)
0.1	156	100	158	100
0.05	152	100	170	100
0.025	176	100	160	95
0.0125	161	85	153	75
0.010	175	62	165	50
Check (1 cc of ethanol)	160	4		

the laboratory-prepared DDT is at least as toxic to mosquito larvae as is the commercial DDT. At dilutions of 0.05 ppm both samples gave 100% dead larvae. In dilutions of 0.01 ppm the commercial sample gave 50% kill, as compared with 62% kill for the laboratory sample.

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Radiocardiography: A New Method for Studying the Blood Flow Through the Chambers of the Heart in Human Beings

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The passage of radioactive substances through the cardiac chambers can now be graphically recorded with the aid of a specially constructed, ink-writing Geiger-Müller counter. This procedure makes it possible to investigate in human beings certain hemodynamic functions not previously accessible to study. Briefly, the method consists of placing a carefully shielded Geiger-Müller tube over the precordium, rapidly injecting 0.1–0.2 mc of radiosodium (Na^{24}) into one of the antecubital veins, and recording the counts by means of the newly devised direct-writing counter.[2] The curve records the concentration of radiosodium in the structures underlying the tube, as represented by the number of disintegrations of the radioactive element per unit of time. The curve is corrected for the random bursts of radiosodium by taking the mean of the counts over a half-second period. Na^{24} has a short half-life (14.8 hrs) and is rapidly eliminated from the kidneys. The quantity injected is within the safe range recommended by the authorities, and the amount of radiation is much less than that which the patient receives during various diagnostic X-ray examinations. In the last year and a half, more than 250 subjects were given injections for radiocardiography without untoward effects.

The reconstructed precordial tracings in normal subjects generally consist of two principal waves connected by a plateau-like transitional zone. The tracings read from right to left, the injection point being at the far right. For purposes of simplicity, the first wave, which traces the blood through the chambers of the right heart, has been called the R-wave, while the second wave may be termed the L-wave, as the left heart receives and expels the labeled blood.

The initial upsweep of the curve is preceded by a level stretch at the base line. This is the period following the injection during which no radioactivity is detected over

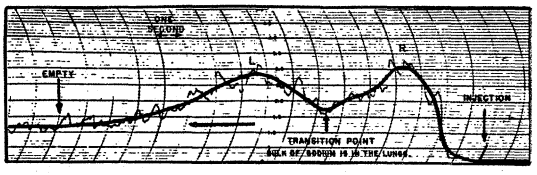

FIG. 1. Radiocardiogram of normal patient. Note characteristic biphasic wave as heart pumps the radioactive blood through its chambers. Each vertical line denotes a 1-sec interval. (Tracing is read from right to left.)

the chest and represents the time interval required for radioactive blood to travel from the site of injection to the precordium. At the termination of this period, the tracing begins to sweep upward to reach the peak of the first wave. This records the entrance of the blood into the superior vena cava, right auricle, and right ventricle. The curve then descends because of the expulsion of the labeled blood from the right heart into the lungs. The line then rises as blood returns from the pulmonary circulation and enters the chambers of the left side of the heart. After the second peak has been reached, the curve descends gradually and ends in a second plateau which lies above the base line. The long descending limb of the L-wave represents the period during which the left cardiac chambers are being emptied of the labeled blood. The end of the L-wave marks the point at which all of the injected radiosodium has completed its passage through the left ventricle. Fig. 1 shows a typical normal tracing.

In some cases the curve consists, not of two well-defined waves, but of a single large wave. The reasons why the curves are sometimes monophasic are as yet imperfectly understood and are being investigated.

[1] Endowed by grants from the Blanche May and Beaumont Research Funds.

[2] We wish to thank Dr. Robert Miller, of Los Angeles, for the design and construction of the apparatus.