through the sliced stems just below the 21-day boll. The treated boll matured 30 days later. The fiber from the treated boll was separated from the seeds and extracted with ethyl alcohol in a Soxhlet extractor for 5 hr (*7*) to remove the waxes quantitatively (*6*). The extracted fiber was air-dried and then boiled in 300 ml of 1% NaOH for 1 hr, the fiber being tied with thread to a glass rod to keep it well under the surface and prevent oxidation. On removal from the alkali, the fiber was rinsed in dilute acetic acid, then in water repeatedly until rinsings no longer were acid, and the sample was dried at 50° C for 2 hr in a circulating-air oven. Yield: 406 mg of purified cellulose.

The purified cellulose was hydrolyzed by the method of Monier-Williams (*8*). A yield of 337 mg of unpurified sugar was recovered from the cellulose hydrolyzate. The crude sugar was made up to 5 ml with water, and a 1/100-ml portion was removed for direct determination of C^{14} activity according to the method of Schwebel, Isbell, and Karabinos (*9*). This procedure indicated that 5.49 μc C^{14} were present in the 5 ml of sugar solution. Thus, approximately 44% of the radioactive C^{14} was converted to cellulose and recovered in the glucose molecule.

The 5 ml (less the 1/100 ml used for analysis) of sugar solution were passed through approximately 5 g of Darco G-60 and Celite according to the method of Whistler and Durso (*10*) and washed with water (10 times the volume of adsorbent) to recover the glucose. This procedure separates the monosaccharides from the disaccharides, the monosaccharides coming off in the water wash and the disaccharides being eluted with 20% ethanol. This separation indicated 95% of the cellulose had been hydrolyzed to glucose by the Monier-Williams method.

The water wash solution of glucose from the carbon-Celite column was freeze-dried and crystallized from methyl and isopropyl alcohol. A yield of 298 mg of crystallized glucose was recovered with a specific activity of 0.0192 μc/mg or 3.459 μc/mM as determined by the vibrating reed electrometer. A 75-mg sample of the radioactive glucose was oxidized by oxygen in aqueous potassium hydroxide[4] to remove carbon 1. The resulting potassium D-arabonate was separated and recrystallized. A 5.82-mg sample of the purified potassium D-arabonate was oxidized and the activity of the resulting CO_2 determined by use of a vibrating reed electrometer. The drift rate was equivalent to 1.1 dps for the 5.82 mg sample. This corresponds to 1.04×10^{-3} μc/mM. Dividing 1.04×10^{-3} by 3.459 (activity of original glucose from cotton) $\times 100 = 0.03\%$. Hence 99.97% of the activity was in carbon 1 of the glucose molecule.

These data are most interesting, since they constitute strong evidence that the glucose-1-C^{14} was synthesized to cellulose directly by an enzyme system of the cotton boll. No one has previously traced the synthesis of cellulose in the cotton boll through the introduction of glucose-1-C^{14} and thus there are no prevailing theories as to the mechanism involved in cellulose synthesis. From the above data one might theorize that the glucose is polymerized directly, possibly receiving the essential energy through phosphorylation.

Later publications will compare the data obtained on the biosynthesis of C^{14}-specifically labeled cellulose in the cotton boll and on that of the cellulose produced by *A. xylinum*.

[4] An unpublished procedure of H. S. Isbell and associates of the National Bureau of Standards.

References

1. Isbell, H. S., *et al. J. Research Natl. Bur. Standards*, **48**, 163 (1952).
2. Isbell, H. S., and Karabinos, J. V. *Ibid.*, 438.
3. Greathouse, G. A., *et al.* The Biosynthesis of C^{14}-Specifically Labeled Cellulose by *Acetobacter xylinum*. Abst., 2nd Biochem. Congr., Paris (1952).
4. Minor, F. W., *et al.* The Biosynthesis of C^{14}-Labeled Cellulose by *Acetobacter xylinum*. Abst., 12th Intern. Congr. Pure Appl. Chem., N. Y. (Sept. 1951).
5. Anderson, D. B., and Kerr, T. *Ind. Eng. Chem.*, **30**, 48 (1938).
6. Marsh, P. B., *et al. Textile Research J.*, **20**, (5), 288 (1950).
7. Conrad, C. M. *Ind. Eng. Chem., Anal. Ed.*, **16**, 745 (1944).
8. Monier-Williams, G. W. *J. Chem. Soc.*, **119**, Part 1, 803 (1921).
9. Schwebel, A., Isbell, H. S., and Karabinos, J. V. *Science*, **113**, 465 (1951).
10. Whistler, R. L., and Durso, D. F. *J. Am. Chem. Soc.*, **72**, 677 (1950).

Manuscript received January 16, 1953.

A Cage for Rearing Predator-Prey Populations of Mites[1]

Thomas Burnett

Dominion Parasite Laboratory, Belleville, Ontario, Canada

At the Belleville laboratory, studies are in progress on the establishment of a predator-prey system, the grain mite *Acarus siro* L. being used as the prey species and other mites as predators. The initial phase of this work dealt with the development of a universe that would provide the following requirements for the studies: (*a*) the mites should develop in as natural a state as laboratory conditions would allow; (*b*) the populations of predator and prey should be spread in a single plane so that most of the individuals could be observed and counted; (*c*) it should be possible to record the distribution of the predator and prey populations; (*d*) physical factors such as temperature, humidity, and the surface over which the predator searches for prey should be regulated; (*e*) if necessary, it should be possible to add fresh food as required; and (*f*) it should be possible to replicate each experiment. A description of a cage designed to satisfy these conditions forms the basis of this note.

The cage consists of 2 square sheets of glass held ½ in. apart by strips of acrylic resin plastic on the four sides (Fig. 1). The base of the cage consists of a sheet of double-diamond glass with smoothed edges.

[1] Contribution No. 3013, Division of Entomology, Science Service, Department of Agriculture, Ottawa, Canada.

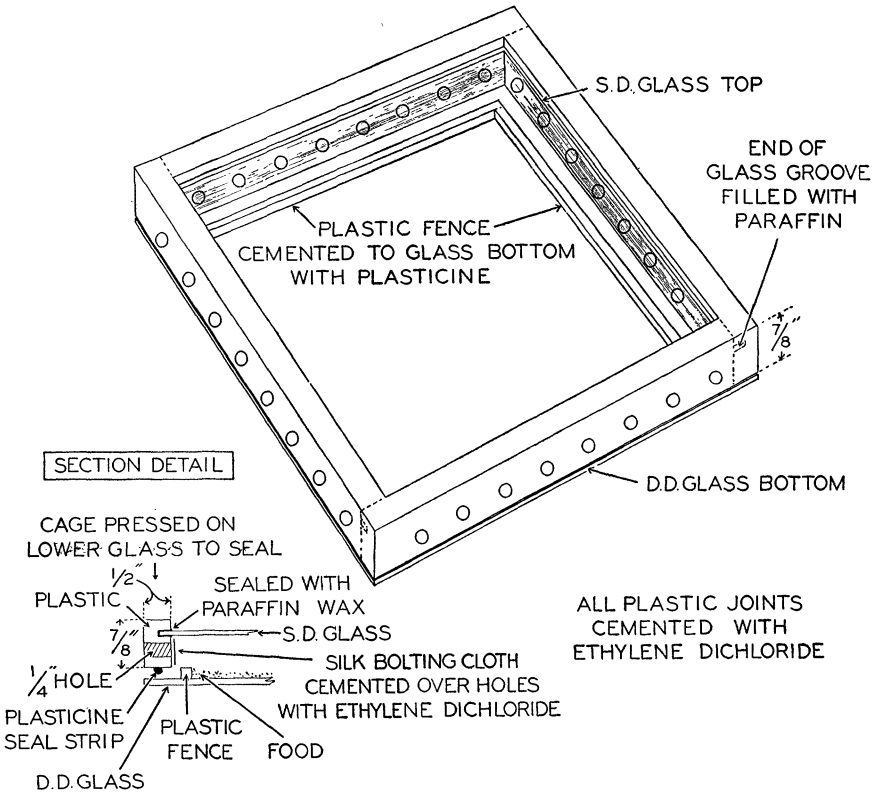

FIG. 1. Cage, with details of construction.

A plastic fence that retains the food is fastened upon the glass plate with plasticine. The sides of the cage are a unit with the glass top. In making the top of the cage the sides are cut from a sheet of plastic ½ in. thick (the length of the sides depends, of course, on the size of the cage), the groove to hold the glass top is cut, and the ventilating holes are drilled. Silk bolting cloth is then cemented, by means of the solvent ethylene dichloride, on the inside of the plastic strips between the groove that holds the glass top and the bottom. For low-density populations of the grain mite, 15 XX cloth is satisfactory, but for high-density populations a cloth of finer mesh is preferable. The four sides are then placed around a sheet of single-diamond glass and the corners cemented together. Finally, the glass is sealed in the groove cut in the plastic with paraffin wax. The top and bottom parts of the cage are sealed by a ribbon of plasticine. The ribbon is formed by forcing black plasticine through a small circular opening in the end of a compression tube similar in design to a hand-operated grease gun.

In practice, a thin layer of food is sifted upon the bottom of the cage and removed from the area outside the plastic fence. Foods that tend to become moldy at high humidities are sprayed with a 2% solution of Shirlan N.A. in 50–70% ethyl alcohol. After the food has completely dried, the top of the cage is sealed in place. Each time individual predators or prey are added or removed from the cage it is necessary to break the plasticine seal. Afterward a fresh ribbon of plasticine is applied. For the observation and census of the mites a binocular dissecting microscope is suspended above the cage and a light source is placed underneath the cage. Movement of the mites can be prevented if necessary either through the use of carbon dioxide or by cooling the cage.

Populations of the flour mite generally move to the edge and corners of the food area when they are introduced into the cage. It is not known whether this indicates a lack of uniformity within the cage or whether it is characteristic of the species. The distribution becomes more uniform a few months later, when the population has increased or has been subject to attack by the predator.

Manuscript received September 22, 1952.

Growth and Regeneration in *Hevea* Seedlings

T. J. Muzik

Federal Experiment Station, U. S. Department of Agriculture, Mayaguez, Puerto Rico

In view of the present interest in Para rubber, *Hevea brasiliensis* (Ex. Adr. Juss.) Muell. Arg., methods of vegetative propagation are of considerable importance. Twigs of mature trees do not form roots, whereas stem