Finally, a series of 8 specimens from different subjects were obtained. The plasma from each was divided into three portions. One portion was tested immediately, and the others were frozen and stored. Eight days later the second portion of each was thawed and the prothrombin activity measured. This process was repeated after a further period of 13 days, when the third portion was examined. The results for the undiluted plasma, given in Table 3, show agreement with those obtained at 25% dilution. It will be noted that there are only minor differences in the average values. Fresh control plasmas studied at the time of each determination showed no significant differences in the thromboplastin solutions used.

TABLE 3

Subject	Prothrombin time (sec)—undiluted plasma		
	Fresh	Stored 8 days	Stored 22 days
A	16.5	17	16.2
B	16.1	15.2	16.3
C	19.3	19.8	19.1
D	18.9	19	18.8
E	19.1	18.3	18.5
F	19.7	19.1	18.9
G	20.6	20.2	...
H	19.7	19.5	...
Average	18.7	18.5	18.0

The first attempts to store plasma by freezing were only partially successful. Evaluation of a number of techniques led to the method finally devised. Four important points in this technique require emphasis:

(1) The samples were kept frozen. Samples accidentally thawed during storage showed a considerable prolongation of the prothrombin time. Freezing in an ordinary "deep freeze" unit was quite satisfactory, and no improvement was noted when samples were rapidly frozen by means of solid carbon dioxide.

(2) Samples to be used in determinations were always thawed rapidly. If allowed to thaw in ice water or in air at room temperature, a faint cloud of precipitated material was often observed in the plasma, and this was usually accompanied by an increase in the prothrombin time. This may be associated with the fibrinogen change reported by others (*5*, *8*). Such phenomena were not observed with rapidly thawed plasma.

(3) Once thawed, determinations were made without delay. Thawed plasma kept at room temperature or at 37° C deteriorated rapidly, the prothrombin time being doubled in 1½ hrs at the latter temperature. Such changes were noted to be similar to those described by others (*7*, *10*) for fresh plasma, but were found to be more pronounced in thawed plasma. Fortunately, when thawed plasma samples were kept in ice water during the period between thawing and determination, no appreciable deterioration occurred for at least 4 hrs.

(4) Storage was most satisfactory when tubes of small lumen were used. The more rapid heat transfer of such tubes presumably aided the necessary rapid thawing of the plasma. The tubes used were easily sealed in a flame, thereby avoiding spillage and loss or contamination. No precautions were taken to avoid bacterial contamination, aside from ordinary cleanliness.

It might also be noted that much of the work was done during hot weather and that this did not demonstrably influence the results.

It is believed that the method described makes possible (1) the use of stored standard plasma samples as controls in the prothrombin test, and (2) storage of plasmas, if desired, until a convenient time for determination is at hand.

References

1. CAMPBELL, H. A., SMITH, W. K., ROBERTS, W. L., and LINK, K. P. *J. biol. Chem.*, 1941, **138**, 1.
2. HONORATO, R. *Amer. J. Physiol.*, 1947, **150**, 381; HONORATO, R., and QUICK, A. J. *Amer. J. Physiol.*, 1947, **150**, 405.
3. HURN, M., BARKER, N. W., and MAGATH, T. B. *J. lab. clin. Med.*, 1945, **30**, 432.
4. KATO, K., PONCHER, H. G., and RICEWASSER, J. C. *Amer. J. clin. Path.* (Tech. Suppl.), 1941, **5**, 110.
5. LOOMIS, E. C., and SEEGERS, W. H. *Amer. J. Physiol.*, 1947, **148**, 563.
6. MANN, F. D., HURN, M., and MAGATH, T. B. *Proc. Soc. exp. Biol. Med.*, 1947, **66**, 33.
7. MARTIN, J. W., CURFMAN, G. H., and CAVANO, F. L. *J. lab. clin. Med.*, 1947, **32**, 554.
8. MORRISON, I. R. *Amer. J. med. Sci.*, 1946, **211**, 325.
9. MUNRO, M. P., and MUNRO, F. L. *Amer. J. Physiol.*, 1947, **150**, 409.
10. PAGE, R. C., and DEBEER, E. J. *Amer. J. med. Sci.*, 1943, **205**, 257.
11. QUICK, A. J. *J.A.M.A.*, 1938, **110**, 1658.
12. QUICK, A. J. *Amer. J. Physiol.*, 1943, **140**, 212.
13. QUICK, A. J. *Amer. J. Physiol.*, 1947, **151**, 63.
14. QUICK, A. J., STANLEY-BROWN, M., and BANCROFT, F. W. *Amer. J. med. Sci.*, 1935, **190**, 501.

An Improved Method of Precipitating and Drying Vinyl Copolymers[1]

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In the course of synthesizing several 1-kg batches of vinyl copolymers there arose the problem of precipitating and drying the large batches in the laboratory.

For most of the copolymers prepared, precipitation in a large vat with methanol gave rise to materials with a molasses-like consistency which were very difficult to dry. This method failed with a number of copolymers including methyl acrylate-maleic anhydride, ethyl acrylate-maleic anhydride, styrene-butyl acrylate (these copolymers were prepared in toluene solution), methyl vinyl ketone-methyl methacrylate, methyl vinyl ketone-butyl methacrylate (prepared in 70% methanol solution), vinyl acetate-vinyl

[1] The work described in this paper was done under a contract, recommended by the Bureau of Aeronautics, between the Navy Department and the Polytechnic Institute of Brooklyn.

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chloride (bulk polymerization), vinyl ethyl ether-acrylonitrile, vinyl *n*-butyl ether-acrylonitrile, and vinyl isobutyl ether-acrylonitrile (prepared in acetone solution).

It seemed that the ideal conditions for precipitating and drying would be those wherein the copolymer would be precipitated with a large surface area, thus expediting complete solvent removal. For the acetone-soluble copolymers (those mentioned above), this was realized in the following manner: The copolymer solution was precipitated in four times its volume of methanol to remove the unreacted monomers. The methanol was then decanted and the copolymer taken up in acetone. The latter was added dropwise or in a very thin stream on a fast stream of water in the laboratory trough. The precipitated copolymer was caught on a screen placed at the end of the trough. This method of precipitation permitted complete removal of the acetone from the copolymer. The latter was then placed on paper toweling (without squeezing out the excess water) and dried for 48 hrs. After this time the copolymers were usually completely dry. If not, final drying was easily accomplished in a vacuum oven at 35° C.

Automatic Masking of Lantern Slides

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In this laboratory we have developed a method for the automatic masking of lantern slides which may prove useful to scientists in many fields. The procedure follows:

The negative is projected to a suitable size, either by enlargement or reduction, upon the easel shown in Fig. 1.

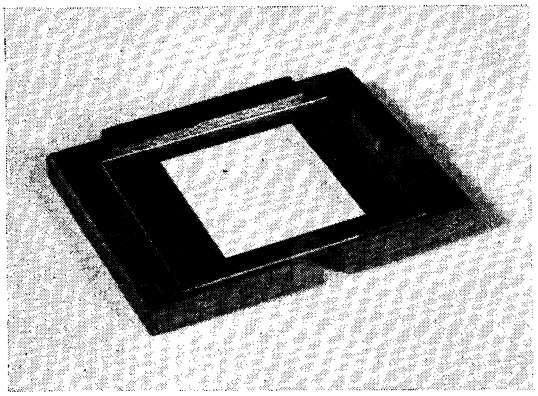

FIG. 1

On the easel is a lantern-slide cover glass, to the lower surface of which a square of white paper, backed with tin foil, has been glued. The subject material is focused and composed on the white paper.

A lantern-slide plate is then substituted and the exposure made in the usual manner. Before the plate is removed, the lantern-slide cover glass is placed on top of it with the tin foil in contact with the emulsion. After removing the negative carrier from the enlarger, the border about the tin foil is then "burned" by double or triple the original exposure time.

On development, there is revealed a lantern slide that has been accurately composed and automatically masked to that composure. Three sizes of composing and masking shields, all of which have 1-cm top and bottom borders cover the range of most subjects. The lateral borders are 1, 2, and 2.5 cm, respectively. Almost any variation is possible. It should be kept in mind, however, that the binding tape accounts for about 0.5 cm, and any masking should exceed that dimension, if for no other reason than to allow space for thumb markers.

A Simple Graphical Solution for Potency Calculations of Multidose Assays

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The necessity for calculation of the results of a number of multidose assays led to search for a simple method of solution. In the tests to be evaluated, samples were assayed at two or three dose levels, conditions varied as to preliminary estimates of potency of unknown, but the number of replicates per sample and standard at each dose were constant in any one experiment. The data from each test could thus easily be handled by the method of Bliss and Marks (*1*), but application of this technic is both tedious and time consuming for routine purposes.

A survey of the literature reveals that Knudsen (*3*) has described a graphical method which might be adapted to the problem in hand. However, her approach is limited by the necessity of drawing a network of radial lines for each dose interval employed as well as for every different assumed potency, and the axes must be rescaled for responses of different orders of magnitude. The nomograph provided for estimation of the error of the assay may be used only where rational basis for grouping of replicates exists; and even if litter mates were used in these tests, there is doubt whether they provide such a basis (*2*).

Sherwood (*4*), on the other hand, has reduced the calculation of potency from such data to relatively simple formulas, and these lend themselves readily to rearrangement which permits simple graphical solution. In the following, the same meanings are to be supplied to the symbols as those described by Sherwood. The case of the two-dose assay will be given in detail; similar reasoning leads to corresponding simplification for the cases of three- and four-dose assays.

The solution of the two-dose assay, as given by Sherwood, is:

$$\% \text{ Potency} = \text{Antilog}\left[2 \pm c + d\,\frac{(U_2+U_1)-(S_2+S_1)}{(U_2+S_2)-(U_1+S_1)}\right]. \quad (1)$$

If the data are substituted in the fraction within the