for the ATP-ase assays. The tissues were homogenized in a Waring Blendor for 5 min and each tissue was assayed at levels of 1 and 2 mg of fresh tissue per assay. The equipment used was kept cold during preparation of the tissues and ice was added during the blending period.

After the tissues were obtained for determination of the ATP-ase activity, the remainder of the carcass was wrapped in moistened paper towels and stored at either −2° C or at +5° C. Samples of tissue were removed 2, 4, 8, or 15 days later and the ATP-ase activity was determined. In some experiments two different samples of muscle tissue were taken from the same carcass on the same day, and the values obtained for the two samples were in good agreement.

TABLE 1

ADENOSINETRIPHOSPHATASE ACTIVITY OF RAT TISSUES AS INFLUENCED BY STORAGE CONDITIONS*

Tissue or organ	Storage temperature	Time of storage (days)				
		0	2	4	8	15
Rat No. 1						
Muscle		14.8	15.0	13.0	15.3	16.2
Kidney		9.9	12.4			
Liver	−2° C	8.1	7.8	6.9	6.4	5.2
Rat No. 3						
Muscle		13.4		12.3	14.4	11.2
Liver		6.5		5.9	5.0	3.3
Rat No. 2						
Muscle		14.1	12.2		15.5	
Kidney	+5° C	12.2	12.8			
Liver		7.5	8.0			
Rat No. 4						
Muscle		15.8		11.9	13.3	

* Values expressed as μg phosphorus liberated per mg of fresh tissue in 15 min at 37° C (*1*).

TABLE 2

ADENOSINETRIPHOSPHATASE ACTIVITY OF BEEF MUSCLE TISSUE AS INFLUENCED BY STORAGE CONDITIONS*

Sample†	Storage temperature	Time of storage (days)			
		0	5	9	15
No. 1		9.3	9.0	8.8	9.0
No. 2	−2° C	7.9	8.1	6.9	7.2
No. 1A	+5° C	7.3	8.5	8.5	8.7
No. 2A		7.3	7.1	7.8	7.2

* Values expressed as μg phosphorus liberated per mg of fresh tissue in 15 min at 37° C.

† Samples 1 and 1A were obtained from the same carcass and were adjacent sections, 1½ in. thick of the *longissimus dorsi* muscle. A similar pairing was made for samples 2 and 2A from another carcass.

The rib-eye muscle (*longissimus dorsi*) from the 11th and 12th rib cut was obtained from steer carcasses that weighed 631 and 637 lb. These samples were obtained 24 hr after the steers were killed and the rib-eye muscle was then dissected free of fat and connective tissue. Two samples were prepared from each carcass; one was stored at −2° C, and the other at +5° C. The samples were approximately 1½ in. thick and were wrapped in paper and stored as indicated. The outer portions of the muscle were removed prior to sampling and 3–4-g samples were taken for ATP-ase assays at 0, 5, 9, and 15 days of storage.

The ATP-ase system was found to be essentially stable in both intact rat and beef muscle tissues during storage for periods up to 15 days with the test conditions used (Tables 1 and 2). The slight increases in activity after storage may have been due to a reduction in water content during storage of the tissues. Results obtained for tissues stored above freezing (approximately +5° C) were essentially similar to those obtained for tissues stored at −2° C in that after 8–15 days no appreciable changes in ATP-ase activity had occurred. Some reduction in the ATP-ase activity occurred in liver as evidenced by the results obtained with tissues stored at −2° C.

Values obtained for rat muscle, kidney, and liver were somewhat lower than those reported by DuBois and Potter (*1*). No comparable data are available for beef tissues. The technique of homogenization used in the present work may not have been as effective in rupturing the cells as that used by these workers. The ATP-ase activity was not increased by increasing the homogenization time to 10 or 15 min. The ATP-ase activity of beef muscle was lower than that obtained for rat muscle.

It is significant, therefore, that the ATP-ase system in these intact tissues is essentially stable for a considerable period after the animal is killed. This is in contrast to the instability of certain other enzyme systems such as the oxalacetic acid oxidase system (*4*).

References

1. DUBOIS, K. P. and POTTER, V. R *J. biol. Chem.*, 1948, **150**, 185.
2. ENGELHARDT, V. A. *Adv. Enzymol.*, 1946, **6**, 147.
3. POTTER, V. R. *Adv. Enzymol.*, 1944, **4**, 201.
4. POTTER, V. R., LEPAGE, G. A., and KLUG, H. L. *J. biol. Chem.*, 1948, **175**, 619.
5. UMBREIT, W. W., BURRIS, R. H., and STAUFFER, J. F. *Manometric techniques and related methods for the study of tissue metabolism.* Minneapolis: Burgess Publishing Co., 1947. P. 97.

A Modern Periodic Chart of Chemical Elements

John D. Clark

252 W. 78th Street, New York City

Since the discovery and elucidation of the actinide series of elements, a reexamination of the perennial problem of the position of the rare earths (and now the actinides) in the periodic chart of chemical elements appears to be desirable. The disposition of these series is far from obvious. Emeléus (*3*) and others have pointed out that the actinides are quite analogous to the lanthanides, but Anderson (*1*) has emphasized their resemblances to the other elements. The accompanying chart (Fig. 1), a development of one published by the author (*2*) in 1933 and recently republished (*5*) in a slightly modified form, but with a much more extensive treatment of the two

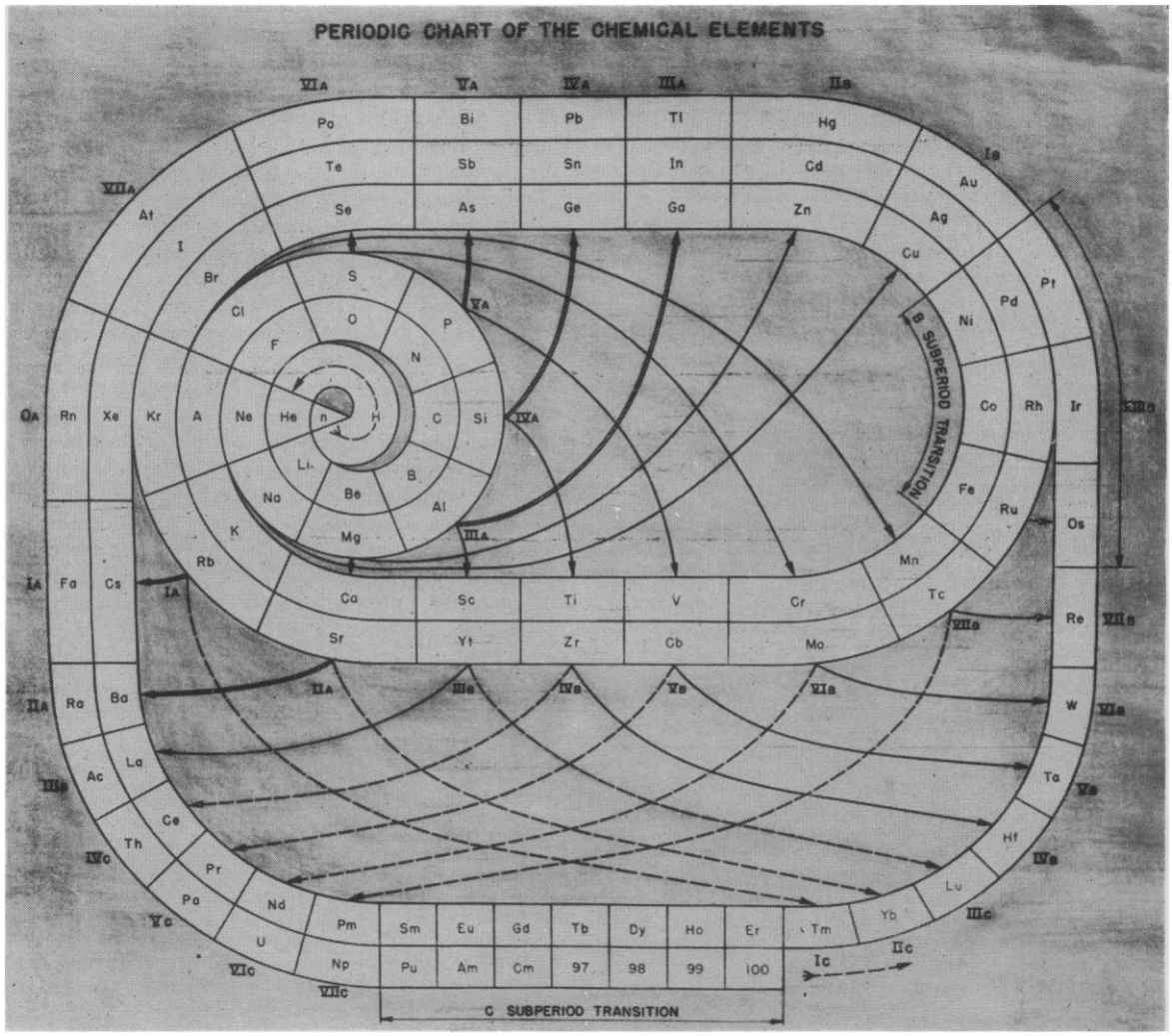

FIG. 1.

series in question, is submitted as a satisfactory solution to the problem.

The most important difference between this and other charts is the division of the groups of elements into three types of groups (instead of the usual division into main and subgroups), resulting from the recognition of three types of subperiods. An A subperiod is that part of a period in which the electron shells of the elements have each been completed by addition of an *s* or *p* electron; the shells of B subperiod elements have each been completed by addition of a *d* electron; and C subperiod elements are those whose shells have been completed by addition of an *f* electron. The A, B, and C groups are defined in the same way. An A subperiod then consists of seven elements plus one transition element (the inert gas); a B subperiod, inserted between IIA and IIIA, has seven elements plus a transition group of the three iron group elements; and a C subperiod, between IIIB and IVB, comprises seven elements and a transition group of seven. Hackh (*4*) pointed out that the consecutive resemblances among elements in a B subperiod approach in importance the periodic relationships, and this phenomenon is even more pronounced in the C subperiods, where resemblances among elements in the series are usually more obvious than similarity to elements in other periods.

The chart presented has the following advantages. It appears to be as simple as possible, consistent with the relationships to be expressed. The idea of continuity is emphasized by the continuous spiral arrangement, and the complex periodicity of the elements is clearly shown. It treats hydrogen logically, as a unique element, putting it at the head of both the IA and VIIA groups. At the same time, it makes a place for the neutron at the beginning of the series and as the first of the inert gases. It separates the A and B groups, but shows their relationships by the tie-lines. (A double tie-line leads to an A group, a single to a B.) The treatment of the lanthanides and actinides is rational and unforced, emphasizing their similarity, but clearly indicating their rela-

tionships to the other elements, (a dotted tie-line leads to a C group) so that such valences as Ce^{+4}, Pr^{+5}, U^{+3}, and Yb^{+2}, appear logically to be expected.

References

1. ANDERSON, J. S. *Nature,* Lond., 1949, **163**, 983.
2. CLARK, J. D. *J. chem. Educ.,* 1933, **10**, 675.
3. EMELÉUS, H. J. *Nature,* Lond., 1949, **163**, 624.
4. HACKH, I. W. *Hackh's chemical dictionary,* 3rd ed. Philadelphia: Blakiston, 1944. P. 629.
5. SEABORG, G. I., MEGGERS, W. F., and CLARK, J. D. *Life,* May 16, 1949, **26**, 82.

Dry Mounts of Amphibian Cleavage Stages

Mychyle W. Johnson

Department of Zoology, Duke University, Durham, North Carolina

Use of dried vertebrate embryological materials for demonstration has recently been reported (*1, 2*). Such methods have been rather widely used but not always reported in the literature.

The use of formalin-preserved amphibian cleavage stages in embryology presents certain difficulties in handling and orientation, especially for students in large classes, and when there is lack of adequate dissecting scopes or light. The method to be described is simple and has proved helpful in the study of amphibian cleavage by classes at Duke University.

FIG. 1.

Formalin-preserved material, previously fixed in Smith's fluid or 10% formalin, is washed in tap water for 2 hr. Jelly and vitelline membranes are then removed by rolling on paper towels. Complete removal of vitelline membranes is important in order to avoid many surface reflections. Bleaching is helpful, and is the next step in the method. Standard techniques such as use of hydrogen peroxide, Javelle water, or hypochlorite solutions are satisfactory. Bleaching is followed with washing in tap water for at least 1 hr and dehydrating through a series of alcohols going from absolute alcohol to xylene, where the material may be stored (or it may be stored in 85% alcohol). The material is put on a paper towel and air-dried for 5 min. It is then ready for mounting or storage.

There are several methods for mounting, but we have found that fastening the material to the tips of paper or clear plastic triangles by means of household cement is successful. The triangles are first pierced with insect pins in a manner similar to that used in gluing small insects. Plastic triangles have the advantage of being usable again in case the specimen breaks.

Some of our dried mounts prepared in this way have been in use for 4 years and show no sign of change. They can be stored in insect boxes while not in use. With a minimum of light and low magnification, the cleavage furrows stand out clearly (Fig. 1). To facilitate handling, the pins can be placed on pieces of balsa or cork when in use. Hemisections of blastulae can be mounted in the same way as cleavage stages.

References

1. JOHNSON, M. W. *J. Elisha Mitchell sci. Soc.,* 1948, **64**. 181.
2. SEDRA, SHOKRALLA N. *Stain Tech.,* 1948, **23**, 205.

Rooting of Haden Mango (*Mangifera indica* L.) Leaf-Bud Cuttings

M. J. Dijkman[1, 2]

University of Miami, Coral Gables, Florida

One of the factors retarding the development of a large scale mango industry in Florida is the lack of knowledge concerning the theoretical aspects of mango propagation and selection. The present paper introduces a new method of propagation which may be helpful in elucidating these problems.

At present, the varieties of mangos grown in this state are propagated by graftage, inarching, and, rarely, by air-layering (marcottage), of scions of known varieties on seedling rootstocks of unknown parentage (*1, 3, 13, 16*). The lack of uniformity in these stocks—some of which are grown from polyembryonic seeds, turpentine, apple, and No. 11 being the commonly used varieties; and others from monoembryonic seeds, such as Haden and Saigon—may be a contributory factor in the tendency toward biennial bearing and other deleterious characteristics apparent in most grafted plants regardless of variety (*1, 3, 16*).

In Florida, the chief emphasis in mango propagation has been on selection of new varieties from chance seed-

[1] Former agronomist of the Society of Central Experiment Stations, and head of the extension service for plantation crops of the South and West Sumatra Syndicate in the Netherlands East Indies (1934–1947).

[2] The author is indebted to Dr. David Fairchild for permission to experiment with his mango trees; to Mrs. Marion Fairchild for helping in editing the paper; to Dr. Taylor R. Alexander, chairman of the Department of Botany, for his cooperation; and to Mr. A. B. Sutton, of the City of Miami Municipal Nurseries, for supplying the coconut husk compost.