Chapter 16

MATURITY METHODOLOGY AND POSTHARVEST PHYSIOLOGY

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A chapter on postharvest physiology and maturity methodologies may seem to combine 2 very diverse aspects of peanut technology; however, the relationship between crop maturity and the biochemical or physiological changes that occur after harvest is of great importance. Although maturity methods are generally regarded as providing information for obtaining maximum dollar value, they also provide the highest overall crop maturity level possible. Relationships of various seed components with maturity are numerous in the literature and generally indicate that at a relatively consistent stage in development, many seed parameters reach a plateau. A crop of seed predominantly in this biochemical or physiological plateau will be of consistently high quality. This quality will be evident in good curing, storage, and shelling characteristics as well as result in peanuts of premium acceptance organoleptically.

This chapter is divided into 2 sections: maturity methodology and postharvest physiology. The maturity section covers all methods found in the literature and deals not only with determining the optimum harvest date for a particular field but also methods which identify arbitrarily defined, sequential maturity stages. These stages can be used to evaluate the seed from a physiological point of view, as well as to determine the specific effect of a given postharvest treatment on peanuts at a particular stage of development. Specific postharvest treatment effects on different maturity stages are important because harvested lots contain many maturity stages. Johnson et al. (1976) found that maturity, as determined by AMI, decreased as seed size (commercial grades of virginia type peanuts) increased. Davidson et al. (1978) and Wil liams et al. (1978) reported that peanut seed size of a given lot of peanuts fits a logistic distribution which shifts with peanut crop harvest date, indicating a high correlation of seed size with maturity. Various quality factors and maturity have been related (Sanders unpublished data), and it is likely that future research will provide defined relationships between maturity, postharvest physiological changes, and quality.

MATURITY METHODOLOGY

The indeterminate fruiting characteristic of peanuts results in seed of varying maturity on the plant as harvest time approaches. The proper time to harvest is when the greatest weight of sound mature kernels are available, since this affects the dollar value of the crop and relates directly to overall high quality peanuts. Methods to determine the proper time to dig peanuts have been used for many years and range from digging a certain number of days after

planting to highly sophisticated techniques requiring equipment valued at thousands of dollars. The information resulting from the methods ranges from a dig—wait-to-dig evaluation to a prediction of the optimum harvest date 2-3 weeks before that time. Under a given set of environmental conditions, probably all methods have some value; however, some methods, such as days after planting and oil color, are so significantly affected by environmental conditions that their general use is precluded. It should be stressed that maturity methods which help determine the best time to dig peanuts are only tools. They must be used in conjunction with frequent field observations since there are conditions of disease, weather, and labor and equipment management that can override any prediction or indication of when the crop is ready to harvest.

In a discussion of methods to determine peanut crop maturity, there are basically variations on only 4 approaches: indirect methods (days after planting, heat units), some relative color evaluation (internal hull color, oil color, methanol extract, pod maturity profile), weight and weight relationships (kernel density, seed hull ratio), and quantitation of a specific component (arginine maturity index). The following is an overview of the methods with details, comparisons, and comments on current and potential usage.

Indirect Methods

Days after Planting. Under optimum conditions a particular peanut variety requires a certain number of days to produce maximum yield. In this method, peanuts are harvested a preestablished length of time after planting. The method is short-sighted and tends to lower crop yield and value, since environmental factors which greatly influence peanut production are disregarded.

The wide variation in time to obtain maximum yield was recently demonstrated by Sanders et al. (1980b). They found that planting dates of 5/1/78, 5/12/78, 5/22/78, and 6/1/78 required, respectively, 150, 139, 129, and 119 days after planting to reach maximum yield in Georgia. In the previous year peanuts planted on 4/29/77 required 153 days to reach maximum yield.

Heat Unit System. Mills (1964) devised a formula based on daily maximum and minimum temperatures to compute effective daily heat units. He then calculated the total heat units necessary from planting to optimum digging date for the years 1958, 1959, and 1960. The 3-year mean indicated that approximately 1600 effective heat units were required for the NC 2 cultivar, a virginia bunch peanut, to develop seed to the optimum stage for harvesting. Mills (1964) suggested that the maturity-heat unit relationship would be valid for similar soils, but that other cultivars would undoubtedly require different numbers of effective heat units. No other testing or use of this method was found in the literature; however, the indirect nature of the method lends itself to error because environmental factors, other than temperature, are not considered.

Effective Langleys Index. Valli (1966) examined 5 heat- and radiation-based meteorological indices for ability to predict peanut maturity. Based on the standard deviation in days and the coefficient of variation, the Effective Langleys index (Langleys times the daily mean temperature) was the best single predictor of peanut maturity. Accumulations of Effective Langleys, from

planting to maturity, varied for the 6 varieties tested.

Relative Color Evaluations

Internal Hull Color. Possibly the oldest method to determine peanut maturity is based on the fact that as peanuts mature the internal surface color of the hull in most varieties changes gradually from white to dark brown, and the seed coat changes from white to dark pink. A maturity determination according to the Shellout Method, as it is generally called, is made on all but the obviously immature (soft, watery) peanuts picked from several plants selected from various field locations. The pods are cracked or cut open, and the percentage of pods with tan to brown coloration inside the hull and pink to dark pink seed coats is determined. There is some variation in the recommended percentage at which harvest should begin. The range is generally 60-80% depending on variety and environmental factors (Bond, 1972, 1975; Henning and McGill, 1974; Allen and Person, 1975; Weete et al., 1979). Few data are available in this area; however, Sanders et al. (1980b) reported that a shellout percentage of $> 75\%$ corresponded to the initiation of the high yield period for 4 different planting dates in Georgia in 1978. In some instances a 60% shellout cannot be achieved because of irregular patterns of pod set; therefore, experienced fieldmen often resort to 3 divisional separations of immature, intermediate, and mature pods as a basis for a judgmental decision on time of harvest. The use of any percentage should be related to condition of the plants, since healthy plants will maintain pods 10 days to 2 weeks longer than plants with severe leafspot (McGill and Henning, 1974).

Miller and Burns (1971) correlated an objective hull color determination with kernel density and light transmittance of oil over 4 days of curing. The objective evaluation demonstrated highly significant differences in internal hull color in subjectively separated mature and immature pods. Gilman and Smith (1977) evaluated 10 peanut genotypes to establish parameters for making reliable maturity determinations on the basis of internal pericarp (hull) color. They found that shellout % obtained from 50 peanut fruits was as reliable in estimating maturity as were kernel density and arginine maturity index.

The shellout method is widely used to estimate the best time to dig peanuts. It can be accomplished in the field with immediate answers, no subsequent sample handling and no equipment. When used conscientiously as a dig—wait-to-dig evaluation, it provides an acceptable degree of accuracy and, if the % mature pods is below that necessary to indicate immediate digging, a careful prediction based on weather, disease management, maturation rate, etc., will be of value. Any estimate, however, should be verified by another shellout evaluation.

Oil Color. Sharon (1963) and Holley and Young (1963) indicated that oil changed to a lighter color as peanuts mature. Emery et al. (1966) and Emery and Gupton (1968) proposed the use of oil color as a maturity index. The index was not designed to be a method for determining when to dig, but a way to evaluate maturity in various lots of peanuts. They indicated that variations could be expected due to the loss of oil color during curing. Holley and Young (1963) reported that oil from slowly cured seed was lighter than oil from rapidly cured seed. Sharon (1963) indicated that carotene was responsible for oil color, and Pattee and Purcell (1967) isolated and identified several carotenoid compounds in peanut oil. Pattee et al. (1969a) reported that the decrease in color with maturity resulted from the dilution of pigments by a rapid increase in oil. In curing studies, Pattee et al. (1968) found decreases in color and suggested an enzyme degradation process involving the enzyme lipoxygenase.

In the spectrophotometric maturity method (APREA Quality Committee, 1971), the density of yellow pigmentation in freshly pressed and filtered raw peanut oil is measured at 450 nm and corrected for incidence of haze by formula application of densities measured at 380 and 520 nm. The validity of the method may be modified by the presence of other influences upon color intensity such as cultivar, length of time in windrow, speed and temperature of curing, and environmental factors such as drought stress which result in deviations from the normal maturation process. General use of this method is precluded, not by the lack of change in oil color, but by the large number of factors that influence that color. The development of a standardized method is highly improbable, since even field to field variation cannot be controlled.

Methanolic Extract. The color of pigments extracted in methanol is the basis for an objective determination of peanut crop maturity resulting in a prediction of the optimum time to dig the crop. The objective nature and predicting feature of the methanolic extract method make it unique. In the method as described for the Florunner variety (Pearson et al., 1973; Holaday et al., 1976, 1979), all peanuts (peg swellings to obviously mature fruit) are removed from freshly dug plants, washed, towel dried, and ground in methanol (2 mL per gram). The mixture is cooled 15 min in a refrigerator and filtered. Light transmittance of the filtrate is determined at 450 nm after 100% calibration with methanol. This value may then be compared with a chart value (Holaday et al., 1979) to predict maturity or simply inserted into the following formula derived by Sanders et al. (1980b): days to harvest = 30-2(75-X), where X = % light transmittance of the methanolic extract at 450 nm. The formula may be further simplified to: days to harvest = 2X-120, and has been tested with only the Florunner variety.

Holaday et al. (1979) suggested that the first maturity determination be made approximately 140 days after planting and another on the date predicted. If % light transmittance on the predicted harvest date is 60 or lower, the crop should be dug. If the % light transmittance is in the low 60's, the date of harvest should be recalculated. However, if at 140 days the light transmittance is 70 or above, the peanuts very likely will not mature normally and predictions based on such data will be inaccurate (Holaday et al., 1979).

In a comparison of the methanolic extract and 3 other maturity methods, Sanders and Williams (1978) found that under optimum conditions the methanolic extract provided an adequate prediction of optimum harvest date but generally did not reinforce the prediction on a sample taken on the initially predicted date. Sanders et al. (1980b) found, however, that drought stress conditions in South Georgia somewhat complicate predictions made with this method. Schubert and Pohler (1978) obtained relatively ambiguous results with the methanolic extract in a comparison of maturity methods in Texas. The nature of the ambiguity was that successive sampling dates predicted ap-

proximately the same length of time remaining until optimum harvest time.

Pod Maturity Profile. Drexler and Williams (1979) described a method for classifying the maturity of individual, fresh Florunner pods which is based on the color and structure of the mesocarp. By scraping or otherwise removing a portion of the pod exocarp, various colors and structural characteristics of the mesocarp are exposed which are directly related to pod maturity. The method of preparing the pods for classification is essentially nondestructive since removal of only the exocarp leaves the remaining pod structure and enclosed seeds intact.

The system of classification may be used to determine the pod maturity distribution at any stage of plant development. Determination of total maturity distribution provides not only the present status of the crop, but also an indication of the number of pods with a potential to mature by harvest date and also represents the overall risk of pod loss relative to pod weight gain. This method is discussed in detail later as a physiological maturity method.

Weight and Weight Relationships

Kernel Density. Although no defined maturity method has been developed using kernel density, there are several reports which correlate maturity and density. Kramer et al. (1963) separated peanuts for maturity on the basis of whether or not they floated in water and found differences in optical density of the oil from mature and immature fruits. A controlled airstream was used by Aristizabal et al. (1969) to separate peanuts into mature and immature categories. They reported that the mean density of immature and mature spanish peanuts from 7 different curing methods were significantly different and possessed considerably different chemical, physical, and organoleptic attributes. Miller and Burns (1971) found that density of immature and mature kernels varied considerably during curing. As the kernels dried, the density of the immature kernels increased sharply and then declined slightly while the lower density of the mature kernels remained fairly constant. An inverse relationship between maturity and kernel density was reported by Gilman and Smith (1977) after they separated pods into immature, intermediate, and mature classes using internal pericarp color.

Kernel Weight. In a 2-year study in Australia, Barrs (1962) concluded that mean individual kernel weight (MIKW) can be used to estimate maturity in peanuts. The method is to determine MIKW on successive occasions until it reaches a constant value, when the crop is mature. The constant, which is apparently a function of variety, was 0.35 g for Natal Common and 0.39 g for Coriante at Katherine, Australia.

Seed/Hull Ratio Maturity Index (SHMI). The SHMI is based upon the changing weight relationship of peanut seeds and hulls during maturation. Pattee et al. (1976, 1977) found that the ratio of seeds to hulls, in either a fresh or dried condition was highly correlated with the various developmental stages of the Physiological Maturity Index (Pattee et al., 1974a) which will be described later. The fresh and dried seed/hull ratios were called fresh weight seed/hull maturity index (FMI) and air-dried seed/hull maturity index (DMI). DMI and SHMI are used interchangeably hereafter.

Pattee et al. (1976, 1977) reported that DMI values for 9 planting and 8 harvest dates over a 2-year period in North Carolina showed that DMI could be applied to estimate average peanut seed maturity under field conditions since the correlation between DMI and yield ranged from 0.88 to 0.98. In 1974 and 1975, they found that maximum DMI for Florigiant was 3.19 and 3.02, respectively, while Florunner had 4.09 and 4.39 maximum DMI. Maximum DMI does not necessarily correspond to maximum yield (Sanders and Williams, 1978), but the difference in maximum DMI between varieties indicated that a SHMI range may be necessary for different cultivar types (Pattee et al., 1978).

Sanders and Williams (1978), in a progress report on studies to compare 4 maturity methods in Georgia, found that a DMI of ca. 3.7 corresponded to the maximum yield period for Florunner peanuts. DMI increased beyond 3.7, but yield decreased. The 3.7 DMI value was in relative agreement with that determined for Florunner peanuts grown for similar periods of time in North Carolina, although no yield data was reported (Pattee et al., 1977). Schubert and Pohler (1978) found that DMI values increased with sampling date in Texas, but the fact that yield did not reach a peak and then decline precluded a SHMI vs maximum yield evaluation. Environmental conditions in Texas as well as North Carolina are sometimes the basis of harvest time since inclement weather may predominate before maximum yields are attained.

Sanders et al. (1980b) found that a SHMI of ca. 3.7 coincided with initiation of the high yield period for 4 different planting dates in Georgia. Under the conditions of that study, which included late-season drought stress, the SHMI was more reproducible and consistent than the shellout method, the methanolic extract, or arginine maturity index.

In the original SHMI procedure (Pattee et al., 1976, 1978; Sanders and Williams, 1978), all the pods from plants selected for evaluation are removed, washed, and towel dried. The pods are opened and those at or beyond the maturity stage characterized by cracks in the white internal pericarp are separated into seed and hull. Less mature pods with no such cracks are placed with the hulls, since in the immature pods, seed weight is negligible. A fresh weight ratio (FMI) of seed weight divided by hull weight is determined; then, the seed and hull groupings are forced-air dried at room temperature for 7 days and a dry weight ratio (DMI) determined. Schubert and Pohler (1978) and Pattee et al. (1978) noted that results with FMI were more erratic than DMI when samples were not handled to avoid moisture loss before analysis. Sanders and Williams (1978) used small plastic bags to avoid moisture loss from samples and found good correlation between FMI and DMI values. Pattee et al. (1980) compared the average coefficient of variation (C.V.) of FMI and DMI values for 1977 and 1978 and found greater variability among FMI values than among DMI values. The observed differences between FMI and DMI suggest that DMI is the method of choice for general usage. The 7-day air drying time initially proposed for DMI is an unacceptable length of time for a maturity determination; therefore, Pattee et al. (1980) evaluated several drying regimes and determined that drying at 135-150 C for 5-6 hours was acceptable.

The SHMI procedure as recommended by Pattee et al. (1980) is as follows: randomly select 18 plants from a uniform plot area up to 0.81 ha (2 acres) and hand-harvest. If soil is tight and dry, use a shovel or other implement to assist in removing plants from the soil, thus insuring minimum stripping of mature

peanuts from the plant. Remove all fruit including small peg swellings from the selected plants and place them in a container large enough to thoroughly mix the bulked batch of peanut fruit. If a divider is available, divide out a 150-fruit sample. If a divider is not available, randomly sample the bulked peanuts with a small container 3 or 4 times to obtain 150 fruit. Place the subsample in a paper bag or wire container and dry for 5-6 hours at 135-150 C. Allow sample to cool for 30 minutes, and then separate seed and hulls except for "raisins" and "pops." ("Raisins" are very immature pods which shrivel upon drying. "Pops" are apparently normal pods which are essentially empty.) Place the raisins and pops with the hulls. Weigh the hull and seed fractions. Divide the seed weight by the hull weight. The SHMI value obtained is indicative of the average maturity level of the peanut fruit in the plot with an average standard deviation of ± 0.2. This procedure is recommended for small plot use and must be extrapolated as to number and location of sample plants for large acreage. Published SHMI values that correspond to maximum yield are: NC-2, NC-5, and Florigiant in North Carolina - 2.9 to 3.1 (Pattee et al., 1980), and Florunner in Georgia - 3.6 to 3.7 (Sanders and Williams, 1978; Sanders et al., 1980b).

The SHMI is well suited for on-farm use since it is simple and no extensive equipment beyond an oven and simple scale is needed. If field data continue to substantiate the results of research on development of this method, the potential for use along with the shellout method is great.

Specific Component Quantitation

Arginine Maturity Index (AMI). One of the greatest criticisms of most maturity methods is their subjective nature. The AMI was developed on objective determinations and, as such, along with the methanolic extract, is unique in that feature. The method is based on the inverse relation between peanut seed free arginine and maturity (Mason et al., 1969; Young and Mason, 1972). This relationship was incorporated into an automated method of determining the arginine content of peanuts and forecasting optimum harvest date (Young, 1973). The method, which relates free arginine and dry matter content as a ratio, is called the Arginine Maturity Index (AMI). During normal pod development, the ratio decreases to a minimum and the time in a particular field when the minimum is reached is correlated with highest yields and time of digging. Although considerable variation may occur, the trend in pod development is usually similar from year to year and, therefore, the changes in AMI values are similar. This facilitates the use of a graph of several years' data to estimate the digging time for AMI values taken in subsequent years. However, the method must be used with the understanding that unusual environmental conditions affect pod development and associated AMI values (Weete et al., 1979; Sanders et al., 1980b).

As with the methanolic extract, the AMI value is determined on all the pods from several plants. A sample size of about 1.5 liter is adequate. The pods are chilled from field to laboratory, where they are washed and prechopped for 1 minute in a food chopper. Dry matter determinations are made by drying 20 g subsamples for 5 hours in a 110 C forced-air oven. Two 30 g samples are each blended for 30 seconds with 200 mL 2% trichloroacetic acid. The solution is poured into a beaker, allowed to stand 10 minutes and filtered. The filtrate is

analyzed in a continuous, automated flow system using the modified Sakaguchi reaction for arginine determination. The colorimetric response is measured at 510 nm and AMI values are calculated on a dry weight basis (Young, 1973). Estimated digging dates are calculated from the equation established for Georgia, $y = 7(X-36)/32$ where y = days to harvest and x = AMI. In the southeast, AMI is normally determined first at about 100 days after planting (or 30-40 days before expected harvest) and again at 14-21 days before harvest, based on the first estimate. The two dates should agree relatively well, and if so, they may be averaged. After the AMI value drops below 100 or a minimum value has been reached, no prediction should be made (Young, personal communication). Sanders et al. (1980b) found in a drought stress year that values below 100 often provided adequate predictions.

Young and Hammons (1974) found that cultivar and harvesting time are factors that affect the AMI. Hartzook et al. (1975) used dry matter and free arginine parameters to evaluate maturation in 8 peanut cultivars. They found clear groupings of AMI values for early- and late-maturing cultivars; however, differences disappeared as plants approached optimum maturity. An inverse relationship between AMI and dry matter was found in all cultivars tested. Tai and Young (1977) used 9 F_2 families from crosses among 6 peanut cultivars and breeding lines to investigate inheritance of dry matter deposition and free arginine as a measure of maturity. They found dry matter to be under multiple gene control, whereas free arginine level seemed to be controlled by 2 major genes with partial dominance for the low arginine character. The results they presented suggest that both dry matter and AMI analyses of low generation populations could be used in peanut breeding to screen for any desirable period of fruit maturation. Hammons et al. (1978) established that fresh fruit-AMI and dry seed-AMI for Spancross, Florunner, and Florigiant had correlation coefficients of more than $r = 0.85$. Both AMI determinations were positively correlated to percent of other kernels and negatively correlated to pod yield, percentage of total sound mature kernels, total kernels, dry matter, and mature seed. Young et al. (1972) and Beuchat et al. (1974) found that various fungicides had an effect on peanut maturation as determined by AMI.

Johnson et al. (1973, 1976) evaluated the AMI, which was developed primarily in Georgia, for use with virginia-type peanuts grown in the North Carolina-Virginia area. They found that AMI values decreased in a manner similar to those in Georgia; however, comparison of data obtained in these experiments with those from similar experiments in Georgia showed a much lower AMI value during normal harvest times for the Georgia peanuts. This work suggested the need for development of a revised data base for AMI determination in the North Carolina-Virginia area (Johnson et al., 1976). Fincher et al. (1980) found somewhat variable AMI values in North Carolina during a 2-year study; however, the data were sufficient to derive a tentative optimum harvest prediction equation for North Carolina.

The AMI method was employed by Johnson et al. (1976) for evaluating maturity and quality of commercial lots of cured shelled peanuts. They found that AMI values from three commercial grades of virginia type peanuts significantly decrease with seed size.

Weete et al. (1979) evaluated the AMI in Alabama in a 2-year study. In research plot situations, they found that AMI was a valuable approach in deter-

mining peanut maturity and estimating harvest dates. In a grower situation, they found that yields and grades from AMI evaluated and harvested fields were not significantly different from the remaining acreage on the farms which were harvested according to growers' judgment. The grower study was complicated by some growers who found it difficult to delay harvest until the forecasted date and some who tended to use AMI information for harvesting all their acreage, rather than just that sampled for AMI analysis. Weete et al. (1979) found that the shellout technique compared favorably with the AMI method, but stated that the advantage of the AMI is that it can estimate the optimum harvest date 2 to 4 weeks prior to that date.

AMI has been included in maturity method comparative tests in Texas. Schubert and Pohler (1978) used Tamnut 74, Florunner, and Florigiant cultivars to evaluate maturity methods and AMI indicated digging dates before the higher yields were reached in all cultivars. The data, determined with AMI curves developed in Georgia, possibly indicate the need for a different AMI data base for Texas as is being developed for North Carolina.

In Georgia (Sanders and Williams, 1978; Sanders et al., 1980b), Florunner was the only cultivar used to compare results from several maturity methods. In 1977, Sanders and Williams (1978) reported that AMI provided an accurate forecast of optimum harvest time. However, as harvest time drew near, AMI predicted harvest dates moved later and later. During the high yield period, the method failed to substantiate that it was time to dig. The main emphasis of the method is as a predictor and the procedure does not call for a determination on the harvest date.

Sanders et al. (1980b) reported that the AMI produced variable results on peanuts planted on 4 different dates in 1978. Results were complicated somewhat by drought stress since some early AMI values were below 100, the low limit value for making predictions in Georgia. The data did indicate, however, a respectable degree of accuracy even after AMI decreased below 100.

Physiological Maturity Methods

Studies on the physiological changes as peanuts mature are complicated, because the maturation process is continuous and not separable into distinct stages. However, to facilitate study of physiological changes during maturation, some separation of defined maturity levels must be made. Each maturity stage necessarily represents a small and slightly overlapping range of physiological characteristics; therefore, investigation of these differences requires a consistently accurate basis of classification. There are 2 major methods to determine relative physiological maturity and one week-by-week description of development. They are all based on internal or external physical and morphological characteristics of the hull, seed coat, and seed.

In a study of composition of developing peanut seed, Pickett (1950) suggested that probably the most reliable and simple method of determining maturity includes a combination of texture of the seed, color of testa, tightness of the kernel in the hull, amount of fleshy material in the hull, change of internal hull color, and appearance of the outside of the hull. Schenk (1961a) tagged aerial pegs of Virginia Bunch 67 and Dixie Spanish varieties and dug the resulting peanuts at weekly intervals. Fifty peanuts were dug and 8 to 10 of the most

mature, large, 2-seeded fruits were selected for analysis, photography, and verbal description. The selection of 8 to 10 pods from the 50 and the fact that data from about 2% of the fruits were rejected after the initial inspection because they were obviously less mature than the remaining fruit provides evidence that all the peanuts did not develop at the same rate. The Dixie Spanish variety developed faster than Virginia Bunch 67. Schenk (1961a) reported changes in fresh and dry weight, crude protein, lipids, free fatty acids, carbohydrates, ash, and enzymes. Although the work reported is detailed on a week-by-week basis, it does not provide defined characteristics relative to physiological maturity stages since it is evident that all peanuts of the same age are not necessarily the same physiologically. Pattee et al. (1970) extended the work of Schenk (1961a) by providing definite physical and morphological characteristics for changes thought to occur on a weekly basis. Pattee et al. (1974a) subsequently found that the characteristics used earlier described physiological stages of maturity rather than a weekly progression of development. Figure 1 shows the seed coat and internal pericarp characteristics used by Pattee et al. (1974a). Stage 13 in Figure 1 is representative of approximately the midpoint of stages 13-15 described by Pattee et al. (1974a).

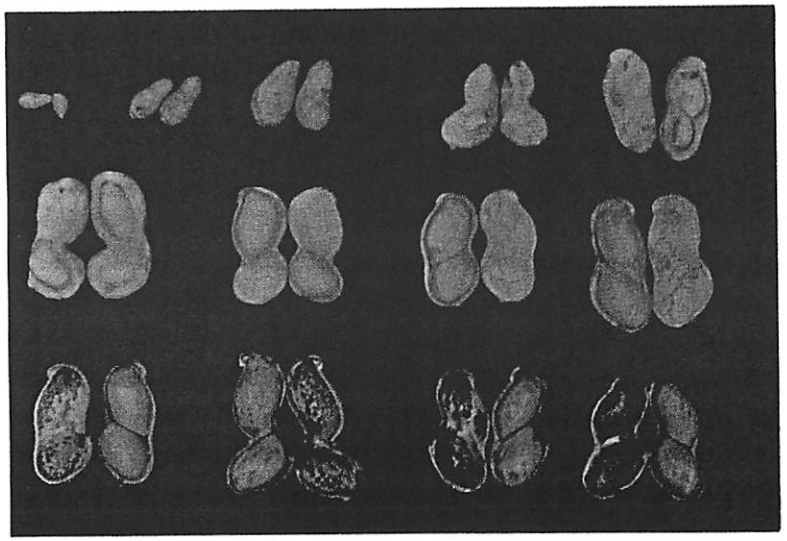

Fig. 1. Physiological maturity index stages of Florunner peanuts grown in Georgia. (Stages 1-13 appear in order from the upper left.)

These characteristics, commonly called the Physiological Maturity Index, have been used to investigate physiological changes in volatiles, lipoxygenase (Pattee et al., 1970); carbohydrates (Pattee et al., 1974a); ^{14}C distribution (Pattee et al., 1974b); lipase (Sanders and Pattee, 1975); tannins (Sanders, 1977); lipid class composition (Sanders, 1980a); fatty acid composition (Sanders, 1980b); and triacylglycerol stereospecific structure (Sanders, 1979). The changes detected in each study indicate a logical physiological progression and verify the accuracy and reproducibility of the Physiological Maturity Index.

Drexler and Williams (1979) developed a method of determining relative pod maturity without cracking open the peanut pod. This characteristic led to

its original description as a non-destructive maturity method, although a small portion of the pod exocarp is scraped away. This method is now called the Pod Maturity Profile. Removal of the exocarp at the point of attachment of the basal seed reveals changes in mesocarp color, veination, texture, and structure which are used to determine the maturity class. Figure 2 shows how combinations of various characteristics relate to the 7 maturity classes.

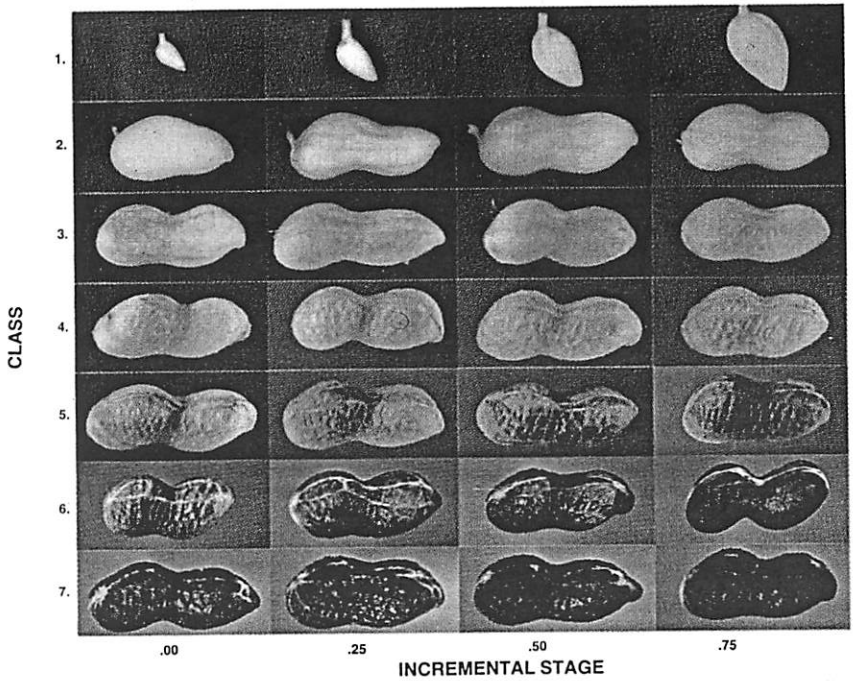

Fig. 2. Florunner mesocarps (exocarp removed) of 7 maturity classes subdivided into increment stages. Reprinted from Peanut Science 8:38 (1981).

Sanders and co-workers (unpublished data) determined oil content, color, free fatty acids, iodine value, and fatty acid profile of each maturity class. As with the Physiological Maturity Index (Pattee et al., 1974a), they found a logical progression of physiological changes which were reproducible throughout the growing season for several planting dates. Although separation of maturity classes is probably no more definitive than the methods of Pattee and coworkers (1974a), there are advantages in the non-destructive nature of the method which indicate its use in specific research situations.

A comparison of the distribution of peanut seed separated by the Physiological Maturity Index and the Pod Maturity Profile is presented in Table 1. The comparison indicates that changes in the external physical and morphological characteristics of the pod mesocarp closely parallel changes in the internal pericarp and testa, since in each Pod Maturity Profile class, over 70% of the pods were placed in no more than 2 physiological maturity stages. A similar range overlap would no doubt be evident in the Physiological Maturity Index if the order of classification were reversed (i.e., Physiological Maturity Index before Pod Maturity Profile).

Table 1. Comparison of Peanut Maturity Classification Methods.[a]

Pod Maturity Profile Class[b]	Physiological Maturity Stage (% in each stage)								
	5	6	7	8	9	10	11	12	13
2	44.8	34.5	13.8	6.9					
3	4.9	15.9	34.9	43.6	0.7				
4			11.4	65.2	23.2				
5			0.1	6.7	44.3	35.8	13.1		
6					0.1	5.0	41.2	45.6	7.2
7							4.9	54.2	40.9

[a]Pod maturity profile classes separated into physiological maturity stages, numbers expressed as % of total. Each value is the mean of 3 replications. (Sanders, unpublished data.)
[b]Each pod maturity profile class contained a minimum of 150 pods.

POSTHARVEST PHYSIOLOGY

Possibly, mistaken ideas that curing is only moisture removal and storage only a matter of maintaining existing quality against environmental factors have led to the sparsity of postharvest research on peanuts. Obviously, many biochemical and/or physiological processes occur during curing and storage and, thus, a wide gap in the peanut information base exists. The postharvest physiology section addresses the general areas of curing and storage work as well as providing special emphasis on the specific areas of flavor and seed dormancy. No attempt has been made to describe all quality changes associated with various curing and/or storage conditions. Instead, there has been an attempt to indicate the spectrum of work accomplished that related physiological processes and various quality indicators.

Curing

Since Dickens and Pattee (1973) presented information on physiological responses to curing in their chapter on "Peanut Curing and Postharvest Physiology," few investigations have been conducted to specifically relate curing to biochemical changes in peanut seed. The following is a synopsis of the information presented earlier in "Peanut: Culture and Uses" plus more recent information.

Respiration. Schenk (1961b) found that maximum respiration rate occurred at curing temperatures of about 42 C and that higher temperatures produced lower rates of respiration and apparent cell damage. In studies on high-temperature—curing-off-flavor, Whitaker and Dickens (1964) found major changes in the respiratory quotient (RQ) for immature peanuts cured at 35 and 52 C but only minor changes in mature seed. RQ was maximum as moisture content of the immature seed neared 25%. This was of special interest as Beasley and Dickens (1963) had earlier found that maximum off-flavor was also produced in peanuts at approximately this moisture content. Whitaker et al. (1974) measured the effects of curing on the internal oxygen concentration of

immature peanuts and found that the minimum partial pressure of oxygen inside peanuts curing at 52 C averaged 4 mm of mercury. This was significantly lower than the average minimum partial pressure of 129 mm of mercury measured inside peanuts curing at 24 C. They concluded that the low oxygen partial pressures found in peanuts curing at 52 C may cause anaerobic respiration which produces off-flavor compounds.

Lipids. Early work on lipids during curing indicated little change in % oil, % free fatty acid, peroxide content or unsaturation level (Bailey et al., 1947; Pickett and Holley, 1960). However, curing treatment does influence oxidative stability as determined by accelerated tests (Pickett and Holley, 1960). Using seed from plants subjected to $^{14}CO_2$ in the field, Mohapatra and Pattee (1973) studied the metabolism of radioactive lipids during curing of the seed at 22 and 50 C. Changes were similar but more pronounced at the higher temperature. Synthesis of radioactive lipid was predominant during the first 6 to 12 hours of curing, whereas degradation dominated between 12 to 24 hours. These changes were related to the moisture content of the curing kernels; a moisture range of 42 to 57% was found to be an important factor influencing the observed pattern of lipid metabolism. Changes in the free fatty acid, monoglyceride, diglyceride, and triglyceride components of a bulk lot of immature peanuts during 50 C curing indicated that both synthesis and degradation of the different components occurred simultaneously. Mohapatra and Pattee (1973) further reported that total lipid content decreased at the same time lipid synthesis increased as evidenced by an increase in lipid radioactivity. Such a situation would be possible if the lipid being degraded was nonradioactive and that being synthesized was radioactive. The data suggest compartmentalization of lipid metabolism in dehydrating peanut kernels. Mohapatra (1970) did not find quantitative changes in the ethanol-soluble constitutents, namely, sugars, organic acids, and amino acids.

Enzymes. Enzyme activity levels in relation to curing have been examined by Pickett and Holley (1960), who found that protease and phosphatase were progressively inactivated by increasing curing temperatures above 49 C. Johns et al. (1968) showed that normal curing enhanced the activity of lipoxidase and suggested that this observation might be related to the loss in color of peanut oil during stackpole curing of peanuts. Sanders (1973) found that lipase was progressively inhibited with time during curing at 50 C.

Flavor

In studying "normal" flavored raw peanuts, Pattee et al. (1969b) isolated and identified pentane, acetaldehyde, methanol, acetone, ethanol, and hexanal as major volatile components and methyl formate, octane, 2-butanone, and pentanal as minor volatile components. The characteristic aroma and flavor of raw peanuts was suggested to arise from a physical interaction of the components isolated and that hexanal was the most significant contributor to this aroma. Brown and co-workers (1971) isolated the aldehydes and ketones from raw peanuts by forming the 2,4-dinitrophenylhydrazone derivatives of these compounds. Concentrations of hexanal and octanal exceeded their flavor threshold values. Based on the concentration data obtained, it was suggested that in addition to hexanal, octanal and possibly nonanal and 2-nonenal contribute to

the "green or beany" flavor of raw peanuts.

Identification of the compounds which form the volatile profile and flavor of raw peanuts provides a base for other studies concerning the mechanics by which these compounds are produced. By following changes in the volatile profile and activity levels of alcohol dehydrogenase and lipoxygenase during maturation, a relationship between the enzymes and their substrates and products, acetaldehyde, ethanol, pentane and hexanal was postulated (Pattee et al., 1970).

Enzymic Origin of Raw Peanut Flavor. The first data suggesting that there might be a direct enzymic role in the production of the raw peanut flavor volatiles is shown in Figure 3. Shortly after the suggestion that pentane and hexanal might be enzymic products of the peanut lipoxygenase reaction within a peanut slurry, Garssen et al. (1971) presented direct evidence that pentane was an enzymic product of the soybean lipoxygenase-linoleic acid reaction. Concurrent with this work, St. Angelo et al. (1972) found only hexanal when they analyzed 100 µl of a peanut lipoxygenase-linoleic acid reaction mixture. Subsequently, Pattee and co-workers (1974c) isolated and purified peanut lipoxygenase and demonstrated that both pentane and hexanal arose from the peanut lipoxygenase-linoleic acid reaction. They further demonstrated that an

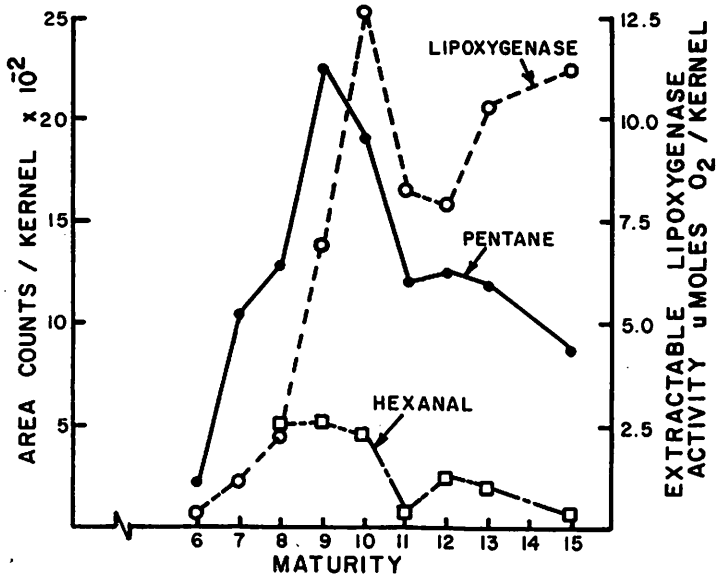

Fig. 3. Relationship of lipoxygenase activity and content of pentane and hexanal in the volatile profiles of peanut kernels during maturation. Reprinted from J. Agric. Food Chem. 18:353 (1970.)

anaerobic condition was not required for the production of both pentane and hexanal, thus distinguishing it from other reported systems. More recently, St. Angelo et al. (1980) confirmed that pentane and hexanal were products found in the headspace of a peanut lipoxygenase-linoleate reaction mixture.

The role of lipoxygenase in the production of flavor volatiles from raw peanuts was further investigated by Singleton et al. (1976). Lipoxygenase was shown to be primarily responsible for production of the flavor volatiles in mod-

el systems of purified peanut lipoxygenase and linoleic acid which produced gas chromatographic profiles almost identical to those of peanut homogenates (Figure 4). The optimum pH for production of the flavor volatiles was shown to be between 6.5 and 7.0. Pentane, but not hexanal, production was inhibited by propyl gallate, hydroquinone, and ascorbic acid. Other factors, such as pH, O_2 content of the reaction system (Figure 5), and temperature have

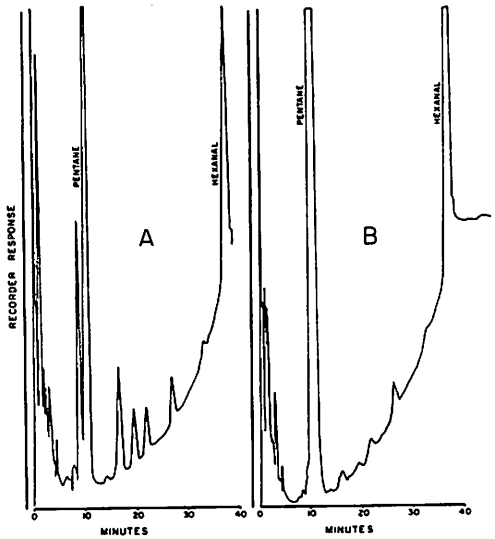

Fig. 4. Typical chromatogram of volatiles from a peanut lipoxygenase-linoleic acid model system (A) and a raw peanut homogenate (B). Reprinted from J. Food Sci. 41:148 (1976.)

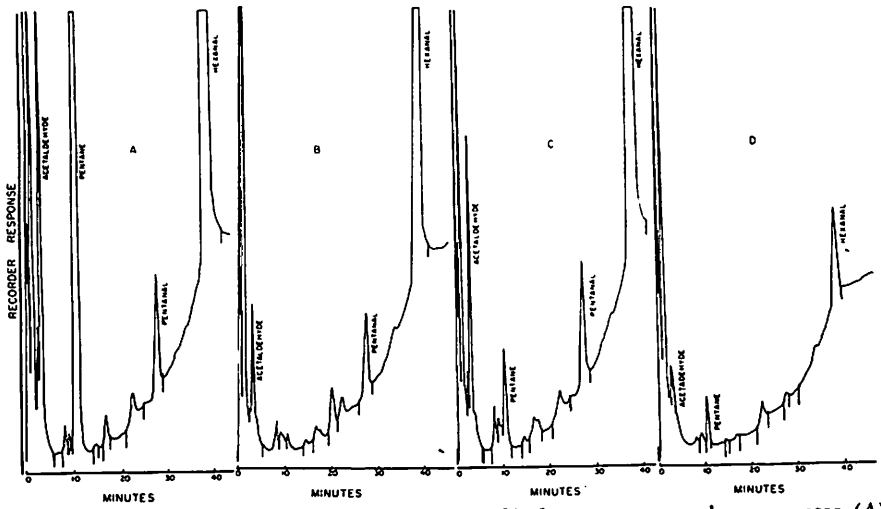

Fig. 5. Effect of O_2 and N_2 gas on the volatile profile from raw peanut homogenates. (A) Blended under O_2 for 60 sec and held at 25 C for 15 min; (B) Blended under O_2 for 60 sec and acidified immediately; (C) Blended under N_2 and then O_2 and acidified; (D) Blended under N_2 atmosphere and acidified. Reprinted from J. Food Sci. 40:386 (1975.)

also been used to differentiate pentane and hexanal production by the peanut lipoxygenase-linoleic system (Singleton et al., 1975).

St. Angelo et al. (1980) examined the aqueous phase of a peanut lipoxygenase-linoleic acid reaction mixture and found only hexanal when the 0.2 mL sample was analyzed by introduction into a 25 C injection port on the gas chromatograph. However, when the spent sample was again introduced into the injection port at 120 C, several compounds, including pentane, were detected. This latter work suggests that pentane arose as a thermal degradation product, as published by Evans et al. (1969). Obviously, pentane may arise as a product of a lipoxygenase mediated enzymic reaction (Garssen et al., 1971; Pattee et al., 1974c; St. Angelo et al., 1980) as well as from the thermal decomposition of aqueous system hydroperoxides during analysis (Evans et al., 1969; St. Angelo et al., 1980). Each reader must evaluate for himself the information presented on lipoxygenase reaction pathways, reaction conditions and products as some differences do exist in published information.

Characteristics of Peanut Lipoxygenase. Siddiqi and Tappel (1957) were among the first to demonstrate the presence of lipoxygenase in peanuts. Using crude extracts and a pH 7.0 assay, they reported peanuts to contain only 1% of the lipoxygenase activity found in soybeans. Dillard et al. (1960), using crude peanut lipoxygenase, found 2 pH optima with linoleic acid, trilinolein, and cottonseed oil substrates. This indicated that more than 1 isozyme of the enzyme might be present. A pH optimum of about 6.0 was reported by St. Angelo and Ory (1972), who used ammonium sulfate to fractionate the enzyme. No activity was present in the partially purified lipoxygenase preparation at pH 8.0. Sanders et al. (1975) reported the separation of 3 peanut lipoxygenase isozymes using DEAE-Sephadex A-50 and a linear gradient of 0.04 to 0.25 M NaCl. Lipoxygenase 1 had a pH optimum at 8.3 while lipoxygenase 2 and 3 had pH optima of 6.2. All 3 isozymes had a molecular weight of 73,000 daltons. Peanut lipoxygenase isozyme (pH 8.3) was strongly activated by 0.5 to 1.0 mM Ca^{++}, and the rate of activation is maximum when the ratio of substrate to Ca^{++} was about 2:1 (Nelson et al., 1977). Peanut lipoxygenase isozymes 2 and 3 (pH optima 6.2) were activated by calcium but did not have an optimum level of activity. Calcium differentially activated peanut lipoxygenase, causing the rate of pentane production to increase more rapidly than the rate of O_2 consumed by the enzyme reaction. At pH 6.2, in the absence of calcium, percentages of the hydroperoxide isomers produced in the peanut lipoxygenase-linoleic acid reaction were 74.9% 13-hydroperoxy *cis*-9, *trans*-11-octadecadienoic acid (13 LOOH cis, trans), 2.6% 13-hydroperoxy *trans*-9, *trans*-11-octadecadienoic acid (13 LOOH *trans, trans)* and 22.5% 9-hydroperoxy 10, 12-octadecadienoic acid (9 LOOH). The presence of 1 mM Ca^{++} at 6.2 did not significantly affect the percentage distribution of the hydroperoxides produced. However, at pH 8.3, the percentage distribution of hydroperoxides produced was 42.2% 13-LOOH *cis, trans,* 10.9% 13-LOOH *trans, trans* and 43.9% 9-LOOH in the absence of Ca^{++} and 57.0% 13-LOOH *cis, trans,* 8.0% 13-LOOH *trans, trans* and 35.0% 9-LOOH in the presence of 1 mM Ca^{++}.

The isolation of the geometrical isomers of hydroperoxyoctadecadienoic acid from the peanut lipoxygenase-linoleic acid reaction (Pattee and Singleton, 1977, 1979) raised the question as to which were enzymically produced and

which were autoxidation products. By taking optical rotation and optical rotatory dispersion measurements of the geometrical isomers, Pattee and Singleton (1979) showed that 13-LOOH *cis, trans* and 9-LOOH *trans, cis* were both enzymic products of the reaction while the *trans-trans* isomers arose by autoxidation routes.

Relationship of Raw Peanut Volatiles to Quality. The findings that the flavor of raw peanuts is almost totally produced by the enzymic reaction of lipoxygenase and linoleic acid and that the flavor profile is generated upon maceration of the peanut (Figures 4 and 5) have a major impact upon our understanding and interpretations of findings relating peanut volatiles to quality. The first work on relating the volatiles isolated from raw peanuts to flavor and quality was conducted by Pattee and co-workers (1964, 1965). They demonstrated that qualitative and quantitative differences existed in the volatiles isolated from control and high-temperature-cured off-flavor raw peanuts. The compounds acetaldehyde, ethanol, and ethyl acetate were found to be related to this quality defect. Singleton et al. (1971) compared the volatile profiles obtained from raw peanuts cured at 22 C, 35 C, 45 C, and 50 C and also showed acetaldehyde, ethanol, and ethyl acetate were related to flavor deterioration. An increase in concentration was detected with each increase in curing temperature. Ethyl acetate was not detected in peanuts cured at 22 C; however, all 3 compounds increased considerably at 50 C. Ethyl acetate was proposed as a specific indicator of the high-temperature-curing quality defect. Using peak ratioing of selected components and organoleptic evaluation data from the samples, Singleton et al. (1971) proposed that this technique of analysis could serve as a basis for objective evaluation of raw peanuts. Too high a temperature during blanching has also been shown to result in an increase in some components and pronounced flavor changes (Pattee and Singleton, 1971). The peanuts blanched at 149 C had initial roasted flavor characteristics which upon storage for periods exceeding 30 days changed to undesirable flavor characteristics.

The volatiles of shelled and unshelled raw peanuts have been shown to change with storage time under simulated warehouse conditions and under controlled environmental conditions of 7 C and 60% R.H.; however, a change with quality has not been established (Pattee et al., 1971). It was speculated that the observed changes could be correlated with an observed "aging period" required to develop maximum roasted flavor potential. The volatile production level reached a maximum between 90-120 days of storage time, and the aging period for maximum roasted flavor potential is considered to be about 3-4 months after harvest. What is being seen is a possible biochemical aging effect occurring during storage which could have a significant effect and correlation to development of maximum roasting flavor potential.

Brown et al. (1972b, 1973) isolated the carbonyl compounds from oil fractions of spanish and runner type peanuts. They found the following values for the total carbonyl, dicarbonyl, ketoglyceride, and monocarbonyl fractions, respectively, 116, 36, 69, and 10 μ mol per 100 g of oil (spanish) and 62, 30, 24, and 8 μ mol per 100 g of oil (runner). They suggested that the higher carbonyl content was a result of differences in linoleate content between peanut types and a longer storage period for the spanish type peanuts (9 months). The higher linoleate content of the spanish type combined with the longer storage period was postulated to allow more autoxidation to occur even though the peanuts were in cold storage. Sanders et al. (1980a) found that total carbonyls increased in peanut grade samples after 3-6 months storage in warehouses with obviously deficient storage conditions. Changes in total carbonyls were concomitant with increases in free fatty acids. The data suggested that the largest changes occurred soon after storage began while ambient air temperatures were high.

A comparison of volatile profiles from 13 different peanut varieties, which sampled the 4 different peanut types, showed no qualitative differences to be present in the profiles (Pattee and Singleton, 1972). However, using peak ratio analysis, several significant quantitative differences were seen. When the 6 varieties from 5 countries (Argentina, Australia, Israel, South Africa, and Taiwan) were compared to those grown in the United States, higher average values for pentane and lower values for methanol and hexanal were found. Pearl, a variety of known poor quality, accounted for 37 % of all significant ratios within the Virginia location where it was grown. From this finding, the authors suggested that a comparison of volatile profiles of new breeding lines of peanuts against the standard aromagram might be utilized to eliminate poor quality lines early in the breeding program, thereby saving considerable time in the development of new commercial varieties (Pattee and Singleton, 1972).

The method of cryogenic-vacuum distillation for volatile concentration does not lend itself to use for routine analysis. A simple, direct gas chromatographic procedure has been developed (Brown et al., 1972a) and used for analyzing volatile components from raw peanuts. Brown et al. (1977) used this method to analyze 5 cultivars grown in North Carolina and Virginia. Peak ratio analysis indicated that the ratio of ethanol-to-methanol and ethanol-to-total volatiles was significantly correlated to taste panel flavor scores after roasting the peanuts. The negative correlation between ethanol and roasted flavor is in agreement with the report of Singleton et al. (1971) on ethanol content and raw peanut flavor. The direct gas chromatographic method was recently modified and improved by Legendre et al. (1979). The modifications include: accommodation of different size samples; obtaining sample volatiles without prior enrichment techniques; providing uniform heating to enhance volatile elution, thereby improving sensitivity; removal of moisture and air to facilitate mass spectral analysis; and a closed system which minimizes loss of low molecular weight volatiles during elution, thus producing a more reliable profile of volatiles. The results suggest that volatile profile analysis may have potential as a quality control method; however, the methodology to implement this knowledge in a time-span that finds practical application has not yet been developed. Future developments in this area shall, without a doubt, overcome this temporary limitation.

Dormancy

Dormancy of peanut seeds is a natural phenomenon which affects several aspects of practical peanut culture. Dormancy can be beneficial when it prevents mature seeds from sprouting before harvest. It can be detrimental when dormancy reduces stand or raises required seeding rates and, therefore, production costs. The commercial seed company must be able to deliver high quality seed

at planting time which contain a minimum of dormant seed. While it is beyond the scope of this section to discuss dormancy in detail, some necessary groundwork will be laid in order to lead up to the main topic: postharvest aspects of dormancy.

Villers (1972) defined dormancy as "the state of arrested development maintained solely by unfavorable environmental conditions such as inadequate water supply." Secondary dormancy is when "dormancy becomes imposed upon seeds by imbibition under conditions unfavorable to the germination of that species, e.g., at temperatures normally favorable for germination of that species until some special release stimulus is applied" (Villers, 1972). For the purpose of this chapter, we will be discussing primary dormancy. It is, however, important—both in research or production settings and in the review of articles and scientific papers—to determine whether one is dealing with primary dormancy, secondary dormancy, quiescence, or loss of viability in nongerminating seeds.

Bailey and Bear (1973a) briefly reviewed the taxonomy of commercial peanuts grown in the USA as it relates to dormancy. *Arachis hypogaea* L. has 2 subspecies of commercial importance: ss. *hypogaea* (virginia type); and ss. *fatigiata* Waldron, which includes var. *vulgaris* Harz (spanish) and var. *fastigiata* (valencia). Despite the generalization that subspecies *hypogaea* has dormancy but subspecies *fastigiata* does not, Bailey and Bear (1973a) found considerable levels of dormancy in some spanish and valencia lines, especially when the peanuts were cured to 5 to 7% moisture in 8 to 16 days. When the lines were cured 4 to 7 weeks in field stacks, the expected lack of dormancy was observed in most spanish and valencia lines. Under the rapid-curing conditions, certain spanish and valencia lines exhibited 70% dormancy while one virginia line had only 3% dormancy. Hull (1937) concluded from inheritance studies that length of dormancy period was determined by multiple factors or quantitative inheritance. Many of the studies which will be cited by Indian authors, e.g. Narasimhareddy and Swamy (1979a), compared Tindivanam Variety-3 (TMV-3)—a dormant, runner peanut line—with TMV-2—a non-dormant, erect type. Some genotypes have dormancy and some do not. The generalization that virginia types exhibit dormancy while spanish and valencia types do not must be recognized as having exceptions; therefore, the trait must be ascertained for each genotype in question.

Developmental Aspects of Dormancy. Seed from dormant and non-dormant peanut genotypes have been found to possess certain biochemical differences, both during seed development and at maturity. Sreeramulu and Rao (1971a) found that auxin patterns were similar during development of dormant (TMV-3) and non-dormant (TMV-2) seed, increasing from 20 to 30 days and declining from 30 days to maturity at 50 days. Auxins apparently were involved in seed growth and not in dormancy. While germination inhibitor concentrations increased in acidic fractions of extracts of both dormant (TMV-3) and non-dormant (TMV-2) seed from 40-50 days, the concentrations were much higher in the dormant seed. Narasimhareddy and Swamy (1979a) reported that dormant (TMV-3) seed accumulated an abscisic acid (ABA)-like inhibitor from 20 to 50 days of development; non-dormant (TMV-2) seed increased ABA-like content from 20 to 40 days, followed by a rapid decline. They also found that dormant seed contained less cytokinins than did the non-

dormant. Ketring and Morgan (1972) showed that exogenous ABA mimics natural dormancy in NC-13 peanuts. Sreeramulu and Rao (1971b) reported that dormant TMV-3 seed had a greater concentration of phenolic acids throughout development than did the non-dormant TMV-2. There were also qualitative differences in phenolic content. The dormant genotype tended to accumulate a higher content of the inhibitory phenolic acids (p-hydroxybenzoic, ferulic, p-coumaric, and vanillic), while the nondormant genotype had a preponderance of synergistic phenolic acids (chlorogenic, caffeic, and protocatechuic). Coumarin content increased during seed development in the dormant cultivar and decreased during development of non-dormant peanut seed. Sreeramulu and Rao (1969) had previously reported that exogenous coumarin caused non-dormant TMV-2 seeds to show signs of dormancy. Gibberellin-content rose rapidly from 20 to 40 days of development in both dormant and non-dormant seed (Sreeramulu and Rao, 1972b). Gibberellins declined markedly from 40 to 50 days in dormant and slightly in non-dormant seed, resulting in low gibberellin content in the dormant and higher gibberellin content in the non-dormant seed. One fraction which co-chromatographed with GA_3 remained constant in nondormant TMV-2, but decreased in dormant TMV-3 seed during development. As will be further discussed later, mature, dormant NC-13 seed exhibited a low ethylene-production capacity (Ketring, 1971) while non-dormant Starr seed had a high ethylene-synthesis capacity (Ketring, 1973).

In summary, mature seed of dormant peanut genotypes had relatively high ABA-like inhibitor content, high phenolic content which were predominately inhibitory in nature, high coumarin content, low cytokinin content, low gibberellin content, and low ethylene-producing capacity. Mature non-dormant peanuts had relatively low ABA content, low phenolic acid content with predominately synergistic types, low coumarin content, high cytokinin content, high gibberellin content, and the capacity to produce at least 2-3 nLethylene per gram fresh weight per hour.

After-Ripening Effects on Dormancy. Hull (1937) defined after-ripening as the process which operates during the required rest period to terminate dormancy caused by some internal physiological condition. As we saw in the previous section, dormant genotypes exhibit physiological traits during development and at maturity which are different from those of non-dormant types. After-ripening would then involve either loss or inactivation of inhibitors present or synthesis of materials which promote germination. Sreeramulu and Rao (1971b) found that coumarin content in the dormant (TMV-3) seed declined to low levels typical of the non-dormant (TMV-2) during after-ripening. In other research involving the dormant genotype TMV-3 (Narasimhareddy and Swamy, 1979b), endogenous ABA-like inhibitors decreased during 40 days of after-ripening at 30-35 C. The decline was slow for the first 10 days after harvest and rapid from 10 to 30 days. By 40 days, when dormancy was absent, ABA content was negligible. Changes in cytokinin activity were less than those for ABA, but increased slightly during after-ripening, peaking at 30 days, and then declined to 40 days. The authors concluded that these hormonal changes are similar to those reported for stratified seed and buds of other species. Sreeramulu and Rao (1972a) also reported a decrease in ABA content during 40 days of after-ripening, at 30-35 C, in initially dormant TMV-3 seed.

During the first 10 days of after-ripening, the fresh weight and respiration, as indicated by O_2 uptake and CO_2 evolution, declined rapidly as the seed dried. Respiration slowly declined from 10 to 20 days and then increased slightly from 20 to 40 days. During the 40-day after-ripening period, dormancy declined from 100% at 0 and 10 days, to 60% at 20 days, to 0% with many stunted seedlings at 30 days, and finally to 0% with normal seedlings at 40 days. Abscorbic acid and starch decreased and both reducing and non-reducing sugars increased during the 40-day after-ripening period.

Among the classical mechanisms of seed dormancy are an impervious or tough seed coat (Hull, 1937) and inhibitors which must be leached out of seed coats or seed before germination can proceed (Toole et al., 1964). Swamy and Narasimhareddy (1977) reported effects of after-ripening on electrolyte leakage from dormant TMV-3 peanuts. Changes in seed coat or membrane permeability would be expected to be expressed in the solutes which leak out of the seed during soaking. As dormancy decreased from essentially 100% to 0% from 0 to 40 days of after-ripening, the quantity of electrolytes leached out of the seed increased. Swamy and Narasimhareddy (1977) cite observations by other scientists that, during dormancy in *Avena atua* L. seed, membrane phospholipid and protein synthesis is high. This suggests that relief of dormancy might involve membrane breakdown or structural change which allows greater seed permeability to water or the leaching of inhibitors. Research findings by Toole et al. (1964) indicate that germination of dormant peanut seed is increased slightly by frequently changing the water used to imbibe seed. This may be due to leaching of some inhibitor (although it is also possible that this treatment only provided more water needed in germination).

Ketring and Morgan (1970) reported that the ability of NC-13 peanuts to synthesize ethylene increased as dormancy decreased over a period of after-ripening. They later expanded their study of this relationship (Ketring and Morgan, 1972), showing that, during a 40-week after-ripening period, ethylene production rates increased from less than 1 to more than 3 nL/g fresh wt per hour as germination increased from 14 and 21% to 60 and 80 + % in basal and apical seed, respectively. They also observed that internal ethylene concentrations of dormant-imbibed, nongerminating peanuts rose as the dormancy level of the entire seed lot declined, indicating that "ethylene production capacity rises before after-ripening is complete."

Dormancy and Postharvest Chemical Treatments. Toole et al. (1964) reported the influence of several postharvest treatments on dormancy in peanuts: (1) puncturing or removing part or all of the seedcoat increased germination of dormant seed; (2) germinating seed in a sealed container increased the % germination; (3) flushing germination boxes with 1% CO_2 for apical seed, and 1 to 10% CO_2, for basal seed, broke dormancy; (4) ethylene applied at 100 ppm induced germination in both mature intact seed and immature seed with seedcoats removed. Removal or partial removal of seedcoats may be related to seedcoat permeability, as previously mentioned concerning increased electrolyte leakage during after-ripening (Swamy and Narasimhareddy, 1977) and effects of leaching seed on germination (Toole et al., 1964). The effect of sealing the germination box is probably due to the buildup of ethylene, or possibly CO_2, in the box.

Bailey and Bear (1971) reported that treatment of dormant runner peanuts with 2-chloroethylphosphonic acid (CEPA or ethephon) stimulated germination. Ethephon is a compound which releases ethylene. Bear and Bailey (1971) observed no increase in germination of virginia peanuts when germinated in closed containers. They theorized that ethylene produced by the seed which germinated should have induced dormant seed in the same container to germinate, as was observed by Toole et al. (1964). Later work by Ketring and Morgan (1972) may explain this inconsistency; they found that an internal ethylene threshold concentration of 2-3 nL/g of fresh tissue per hour was needed to break dormancy in NC-13 virginia type peanuts. The ethylene concentration attained in the sealed boxes (Bear and Bailey, 1971) may have been below the threshold. They also reported that the fungicide tetraethylthiuramdisulfide (Thiram) increased germination of certain dormant peanut seed. Also, Nur and Gasim (1974) reported that a seed-protectant product, which contains Thiram, reduced dormancy of peanut seed. The effect did not appear to be fungicidal, because sterilizing the seed with mercuric chloride did not affect dormancy. They theorize that Thiram may alter the hormonal balance of the peanut seed.

Bailey and Bear (1973b) reported that ethephon (10^{-3} or 10^{-2}M) could be used to break dormancy by treatment at planting time, or as early as 60 days before planting, with no ill effects. They included Thiram with the ethephon, but found no evidence that the fungicide increased ethephon's effectiveness in breaking dormancy, as might have been expected from observations that Thiram decreased dormancy when used alone. They noted that unrefined, technical ethephon was more effective in breaking the dormancy of rapidly cured seed than was a purified ethephon formulation used during 1 year of the research. They concluded it could be commercially practical to break dormancy by spraying seed which were coated with Thiram (or other fungicide) with ethephon solution, resulting in an ethephon-fungicide slurry coating the seed. In another practical application, Clark (1971) described a procedure for testing peanut seed viability, in which he removed the confounding effects of dormancy by treatment with an ethephon formulation (Ethrel).

As we have seen, exogenous ethylene and ethylene-producing compounds are effective in breaking dormancy in peanut seed, and dormancy is accompanied by an inability to synthesize ethylene above a certain threshold rate. The work of Ketring and Morgan has led to a better understanding of the relationship of dormancy and ethylene. They reported (Ketring and Morgan, 1970) that the natural decline in dormancy over a 16-week after-ripening period was accompanied by increased ethylene production. Ethylene, ethephon, and gibberellic acid (GA) increased germination, and GA stimulated ethylene production. The time-courses of germination indicated that GA first induced ethylene production and subsequently germination. The time-course of germination stimulation was the same with ethylene and ethephon. Ketring (1970) reported that kinetin stimulated germination. Ketring and Morgan (1971) showed that another cytokinin—benzylaminopurine—as well as kinetin stimulated germination and ethylene production and that ethylene production was high at 24-48 hours of germination. In contrast, 2,4-dichlorophenoxyacetic acid stimulated ethylene production beginning at 48 hours, but did not cause germination. Treatments stimulating germination caused a rise in CO_2 prod-

uction at about 72 hours. ABA inhibited germination and ethylene production, but did cause increased CO_2 production. ABA mimics natural dormancy and its action is competitively reversed by ethylene and cytokinins (Ketring and Morgan, 1972). This information, coupled with the observation that ethylene production capacity increases both in the seed population and still-dormant seed during after-ripening or natural decline of dormancy, leads to the hypothesis that the loss of dormancy involves the action of a growth regulator(s) on ethylene synthesis which is initiated within 24 hours of inbibition. The control may be through stimulation of ethylene synthesis by cytokinins, gibberellins, or auto-catalysis by ethylene itself and/or by loss of inhibitors such as ABA. Ketring (1975) reported that the action of ethylene apparently involved protein synthesis from stable messenger RNA at the translation level or at the DNA transcription level.

Ketring (1977) proposed a method for breaking dormancy based on the role of ethylene in peanut dormancy and germination. He used 1% (w/w) Ethrel (ethephon) powder mixed with the seed-treatment fungicide. Virginia and runner types responded well to the 1% ethephon (w/w of fungicide), while non-dormant Starr (spanish) peanuts exhibited retarded emergence at this level. Therefore, Ketring (1977) recommended 1% ethephon in the seed treatment fungicide for virginia and runner peanuts and 0.5% for spanish. This method appears to be easily adaptable to the presently common treatment of peanut seeds with powdered fungicidal formulations. The procedures of Bailey and Bear (1973b) and Clark (1971) indicate there would be no problems expected in using ethephon in liquid fungicidal peanut seed treatment, should this process become common. To date, ethephon formulations have not been cleared for this use commercially, but these or similar methods of releasing peanut dormancy remain interesting biologically, if not economically.

Storage Effects on Dormancy. The temperatures and relative humidities to which peanuts are subjected after harvest have a great deal of influence on dormancy and germination. We will restrict the discussion in this section to dormancy; general effects of storage conditions on seed quality and viability are discussed in another section of this chapter. As previously discussed, Bailey and Bear (1973a) reported higher levels of dormancy in both virginia and spanish peanuts which were rapidly cured to 5-7% moisture in 8 to 16 days by circulating air at 21-35 C, than in peanuts stack-cured for 4 to 7 weeks. Bear and Bailey (1973) also observed this effect of curing method on dormancy. They reported that in all but 3 of 28 lines (1 of the 3 was Florunner), 28 to 30 days of storage at 29.4 C broke dormancy of rapidly cured peanuts. However, 19 of the 28 lines required special conditioning at 29.4 C to reduce dormancy to acceptable levels after storage for 90 days at 4.4 C. Again, 1 of these lines was Florunner, which had 10% dormancy after 28 days of conditioning. Florunner responded even more slowly to conditioning at 29.4 C following 150 days of storage at 4.4 C, exhibiting 22% dormancy. Interestingly, Dixie Runner and Early Runner—both of which are closely related to Florunner—exhibited dormancy levels higher than did Florunner following cold storage. Dixie Runner, however, responded quickly to conditioning at 29.4 C, while Early Runner and Florunner responded slowly.

It was a common practice in much of the work of Ketring and Morgan, which we have discussed extensively, to hold seed at 3 C to maintain a stock of dormant seed for experimental use. Bear and Bailey (1973) report considerable variation between crop years; but, in all but 4 lines studied, dormancy would pose a serious problem in seed stored at 4.4 C for 4 to 6 months and then planted immediately. Hull (1937) noted that low-temperature storage prolongs dormancy, while storage at 30 C for 30 days largely eliminates it.

Cold-storage is usually considered to be necessary to maintain the highest quality and viability in peanuts to be used as planting seed. Therefore, a program of warm-temperature conditioning or chemical treatment is often required, especially in virginia and runner varieties, to reduce seed dormancy to acceptable levels.

Storage

Peanut storage is important, both to production agriculture and to product utilization. Seed are usually stored from 6 to 9 months from harvest to planting time. During this storage period, quality must be maintained to allow production of safe, desirable products and seed with high germination potential for the next crop year. Our knowledge of precise physiological changes which occur during storage is limited, but some information is available. In this section, the effects of shelled and unshelled peanut storage on several quality factors and germination are discussed. Further, food quality information related to storage of shelled peanuts may be found in Chapter 17.

Warehouse Storage Effects on Food-Related Quality of Peanuts. Although farmers stock peanuts are normally stored from 3 to 9 months in 3000-5000 metric ton warehouses, physiology research on this aspect of storage is limited. Studies described below measured some aspect of quality and have little relationship to the processes giving rise to the quality indicators. Johnson and Gilliland (1960), using peanuts stored in various type bins, reported that free fatty acids increase with storage and that percentage germination of peanuts after they are removed from storage depends primarily on the moisture of the peanut kernels at the time they were stored, the temperature of the storage environment, and the length of storage period. Davis (1961a, b) found that carbonyls, peroxides, free fatty acids and other quality factors indicated that quality decreased with storage time and higher moisture levels in farm-type bin storage and simulated storage in relative humidity chambers.

Pattee et al. (1971) found that total volatile content of unshelled peanuts stored under simulated warehouse conditions reached a maximum after 90 to 120 days of storage and then declined. They suggested lipoxidase and pectin methyl esterase as enzymes possibly responsible for the production of the volatiles. Sanders et al. (1980a) found that grade factors decreased and free fatty acids and carbonyls increased in peanuts stored in warehouses with obviously deficient storage conditions. Levels of deterioration were related to seed condition, length of storage, excess moisture, high temperature and mechanical damage.

Storage Effects on Germination. The literature reports a wide range of success in maintenance of germinability of peanut seed in storage. These range from maintenance of high levels of germination in inshell peanuts stored under controlled conditions for at least 9 years (Norden, 1975) to almost total loss of

germinability of shelled peanuts in only 6 months (Marzke et al., 1976). The latter case is unusual; Norden (1975) reported that peanut seed generally remain viable for about 2 years in open storage. High seed moisture and relative humidity (RH) are probably most detrimental to viability during storage. Other factors include temperature, atmospheric content, initial seed quality, bagging material, and atmospheric pressure.

Ketring (1971) reported the effects of various storage conditions on shelled, high and low quality Starr peanut seeds. The high quality seed had an initial germination of 81%; the low quality seed lot, which had been damaged by rainfall while in the windrow, had a 30% initial germination. Storage treatments were 21 C/50% RH, 3 C/20% RH, and 3 C/80% RH. Germination and seedling vigor were greatly reduced by 3 C/50% RH storage. This effect was obviously due to the high RH, because 3 C/20% RH maintained germinability and vigor much better. The 3 C/20% RH and 21 C/50% RH storage conditions gave similar results. The low quality seed deteriorated more rapidly than did the high quality seed, with deterioration more rapid at 3 C/80% RH. It should be noted that the cause of the initial damage in the low quality seed was rainfall and high humidity after digging.

Ketring (1973) reported that germination in shelled Starr peanuts, not treated with fungicide, decreased from 100% to 63% after 8 months to 31% after 9 months of storage at 100% RH, at room temperature. Percent vigorous seedlings, defined by seedling growth rates, decreased from 93% to 13% (8 months) to 0 (9 months). At 10% RH, germination was still 100% with 90% vigorous seedlings after 9 months of storage. At 9 months, the 100% RH lot was visibly colonized by fungi; the 10% lot was, therefore, treated with fungicide, divided into 2 lots and storage continued at 10% and 100% RH. Germination began to decline after 7.5 additional months at 100% RH, reaching 44% germination and 0 vigorous seedlings after 11.5 months. At 21 months, germination was still high in the 10% RH treatment although percent vigorous seedlings had declined to 60%.

Clark (1972) stored shelled Starr peanuts under refrigeration (1.7-3 C and 65-70% RH) and in 2 seed warehouses (conditions unspecified). After 6 months, there were no significant differences in germination and emergence among treatments. After 18 months, germination had declined 5% in refrigerated storage and 15% in warehouse storage. Seedling vigor was higher under refrigerated storage.

As was previously stated, Norden (1975) reported successfully storing peanut seed for at least 9 years. His best storage conditions were 2 to 5 C or -4 to -1 C (with no paradichlorobenzene) with a seed moisture content of no more than 6%. At 18 to 21 C, viability remained acceptable for 4 years, then decreased dramatically. Seed with 8 to 11% moisture content deteriorated more rapidly than did those at 2 to 6%. Paradichlorobenzene was harmful in sealed containers and at temperatures low enough to prevent sublimation. During the 9- year period, with proper storage conditions, the virginia seed decreased from 96 to 88% germination, valencias 94 to 88%, and spanish 92 to 73%.

The importance of low humidities during storage of peanuts is a common finding. Bass (1973) reported that peanuts lost viability more rapidly at 21 C/70% RH than they did at 35 C/50% RH, due to the effect of high moisture. At 10 C/50% RH, 10 C/70% RH, and 21 C/50% RH, peanut seed remained viable for at least 5 years. Bass (1973) cites reports that peanuts are relatively responsive to changes in moisture content compared to other seed. This citation is in agreement with the report by Swamy and Rao (1971) in which they compared peanuts with the seed of 2 other crop species. They found that moisture content of peanuts was most responsive to that of the atmosphere. They defined critical moisture content (CMC) of seed as that moisture content which is in equilibrium with 80% RH air. At their CMC, most seed are extremely vulnerable to rapid deterioration. Peanuts exhibited a lower % moisture at their CMC and reached it more rapidly than did the other species examined. This rapidity of reaching their CMC would be expected to make peanut seed more susceptible to degradation during storage at high humidities.

Marzke et al. (1976) studied the influence of storage atmosphere (air, high CO_2, and high N_2) on peanut germination and quality. They found that 12 months of inshell storage at 4.4 C and 26.7 C reduced germination from 84% to 76 and 72%, respectively. As previously stated, shelled peanuts dropped to practically 0 germination at both temperatures in 6 months. This is an unusual case, but graphically illustrates the accelerated deterioration of shelled vs. inshell peanuts commonly observed. Storage atmosphere had little effect on germination compared to temperature, shelling, or length of storage. Neither seed moisture content nor relative humidity were variables in this study. The researchers concluded that high CO_2 or high N_2 storage would be an acceptable alternative to fumigation for insect damage, as far as germination was concerned. They did recommend, as will be more fully discussed in the section on quality, that O_2 concentration should not be allowed to drop below 1 to 2%. Slay et al. (1978) reported that low-oxygen storage (O_2 concentration was not defined in this abstract) of shelled peanuts gave better sanitation, insect control, and handling and required less storage space and energy expenditure than did conventional storage.

There has been interest in vacuum storage of peanuts or evacuation of storage containers followed by partial release of the vacuum with N_2. A trade article (Anonymous, 1978) indicates that cold storage might not be required if peanut seed were stored in commercially-available containers which have a laminated barrier bag which can be evacuated and sealed inside a corrugated box. It is claimed that acceptable germination was retained with virtually constant seed moisture contents. Pearson et al. (1977) reported that germination in peanuts stored in nylon-saran-EVA resin bags which were evacuated and then back-flushed with N_2 was superior to other non-refrigerated treatments tested and equal to the refrigerated control. After 6 months, germination of peanuts stored under vacuum was lower than that of those in which the vacuum was partially relieved with N_2. Clark (1972) noted that polyethylene bags maintained a more constant seed moisture content than did paper or cloth bags.

Germination-Related Physiological Changes during Storage. Ketring (1971) reported no significant differences in soluble and insoluble protein and reducing sugars among seed lots of greatly different initial germination percentages and among storage treatments which had marked effects on germination. Lower initial seed quality and quality loss during storage was reflected in nucleic acid content. In 1973, Ketring reported that decreases in ethylene and CO_2 production by imbibed seed paralleled declines in germination and per-

cent vigorous seedlings. Marked declines in germination and vigor were accompanied by shifts of maximum ethylene production from 24 to 48 hours and maximum CO_2 production from 48 to 72 hours. Ketring and Morgan (1971) showed that the quantity and timing of ethylene production was critical in germination of peanut seed, but that CO_2 production was not without ambiguity in its relationship with germination. Ketring (1973) found no loss in soluble protein until germination, vigor, and ethylene and CO_2 production had declined radically. Extractable RNA content, however, declined during the decline of germination. At 100% RH, extractable RNA was significantly reduced after only 1 month of storage. All indications were that the characteristics of deterioration were the same at 100% and 10% RH, just more rapid at the higher humidity. The relative loss of nucleic acid—specifically extractable RNA—was less than was the relative decrease in germination. This leads to the conclusion that if RNA loss is the (or a major mode) of action in storage damage to germination, the RNA affected must be specific for ethylene production or some other event occurring at germination.

Das et al. (1976) reported a decrease in phenolic compounds in both seed and shells during 9 months of storage. The ratio of inhibitory to synergistic phenolics, however, increased during this storage period, suggesting another possible mechanism for loss of germinability during storage.

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Chapter 17

COMPOSITION, QUALITY, AND FLAVOR OF PEANUTS

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Peanuts (*Arachis hypogaea* L.) are characterized by high oil and protein contents and a low percentage of carbohydrates and ash. The oil from these seed is of high quality, and a large percentage of the world production of peanuts is utilized as an edible oil source. In the USA, generally, about 60% of the production goes into domestic food use, the end products being peanut butter, salted products, confections, and roasting stock. As with many other foods, interest in the composition and chemistry of peanuts is largely a result of their use as human food. Peanuts are continually utilized for preparation of new and improved products; thus, a more complete knowledge of their composition and flavor properties is desirable.

Since an extensive coverage of the literature through 1970 on the physicochemical properties of peanuts was made by Cobb and Johnson (1973), the main thrust of this chapter is to focus primarily on the literature published in the past decade on the composition, quality and flavor of peanuts.

COMPOSITION

Proteins

Crude protein content of whole seed peanuts ranges between 22 and 30% (Altschul, 1964; Pancholy et al., 1978). Total protein can be separated into albumins, arachin, nonarachin or conarachin (Johns and Jones, 1916; Jones and Horn, 1930; Johnson and Shooter, 1950; Johnson et al., 1950; Evans et al., 1962; Johnson and Naismith, 1963; Dawson, 1968, 1971; Daussant et al., 1969a, b; Cherry et al., 1973). The arachin and conarachin fractions were quantified and their properties were determined (Johns and Jones, 1916; Evans et al., 1962; Tombs, 1965; Dawson, 1968,1971; Neucere, 1969, 1974; Ory et al., 1970; Dawson and McIntosh, 1973; Shetty and Rao, 1974). Arachin represents about 63% of the total protein and contains 2 components in the ratio of about 3 to 1 while conarachin represents about 33% of the total protein and contains 2 components in the ratio of about 4 to 1 (Fontaine et al., 1945). The major difference between these 2 globulins was found in their sulfur content, 0.40% for the arachin fraction, and 1.09% for the conarachin fraction (Johns and Jones, 1916). The arachin fraction was rich in threonine and proline and poor in lysine and methionine, while the conarachin fraction was poor in phenylalanine and tyrosine (Kaneko and Ishii, 1978). In addition, the antigenic structure of the major reserve protein, α-arachin, was unchanged by 1 hour heating at 145 C (Ory et al., 1970). This protein component should be intact in peanut products which are heated during processing.