Department of Medicine,
U.P. College of Vet. Sci.
and Animal Husbandry,
Mathura, *July* 21, 1969.

R. C. PATHAK.
S. R. BANSAL.

1. Gill, B. S., *Ann. Trop. Med. Parasit.*, 1964, **58**, 473.
2. —, *J. Camp. Path.*, 1965, **75**, 175.
3. Gray, A. R., *Immunology*. 1961, **4**, 253.
4. Pande, R. and Pathak, R. C., *Ind. J. Exp. Biol.*, 1965, **3**, 244.
5. Ristic, M. and Sibinovic, S., *Am. J. Vet. Res.*, 1964, **25**, 1519.
6. Seed, J. R., *J. Protozool.*, 1963, **10**, 380.
7. Sibinovic, K. H., Ristic, M., Sibinovic, S. and Phillips, T. N., *Am. J. Vet. Res.*, 1965, **26**, 147.
8. Soltys, M. A., *Parasit.*, 1957, **47**, 398.
9. Weitz, B. G. C., *Nature*, London, 1960, **185**, 788.

A LABORATORY TECHNIQUE FOR MASS REARING OF THE TOBACCO CATERPILLAR *SPODOPTERA LITTORALIS* (BOISD.) *PRODENIA LITTURA* (F.) (NOCTUIDAE : LEPIDOPTERA) ON SYNTHETIC DIET

ARTIFICIAL diets for a number of Lepidopteran insects have been developed by various workers.[1] An attempt was made to develop a synthetic diet for *Spodoptera littoralis* (Boisd.) that will facilitate rearing of this insect all through the year in the laboratory.

The composition and various ingredients used for preparing the diet are given below :

Soaked peas (*Pisum sativum* L.) 50 g. ; Yeast tablet (Alembic) 2 g. ; Agar 300 mg. ; Ascorbic acid 500 mg. ; Calcium pantothenate 150 mg. ; Nicotinic acid 100 mg. ; Folic acid 50 mg. ; Pyridoxin HCl 50 mg. ; β-Sitosterol (Sigma grade) 50 mg. ; Methyl paraben (Sigma grade) 50 mg. ; Formalin 40% 10 drops ; Water 80 ml.

To prepare the diet, agar was placed in a beaker containing 50 ml. of water and boiled to dissolve the agar. The soaked seeds, after removal of the seedcoat, were weighed and put into a glass homogenizer and blended in 30 ml. of water with other ingredients. The agar solution was poured into the food mixture with constant stirring. A small quantity of this semi-solid food was poured into a test-tube of $7\frac{1}{2} \times 2\frac{1}{2}$ cm. which has been previously sterilized by heating in an air oven, and set aside to cool under the fan.

Freshly hatched larvæ obtained from a single egg mass were introduced into each tube at the rate of 10 per tube and the tubes were plugged with sterile cotton plugs. Three or more tubes were held together and kept inverted for the first few days as described by Nachiappan.[2] At the end of the 5th day, the larvæ were separated and kept at the rate of two per tube. They were subsequently separated on the 10th day and kept individually in each tube, till they pupated. During these growing periods the tubes were kept horizontally. Fresh food was supplemented once in five days at the time of separating the larvæ as the food had been eaten by them.

The control larvæ were maintained on castor leaf in small jars as described by Eldefrawi *et al.*[4] The rearing of control and test larvæ was carried out at 85° F ($\pm$ 2) in a cabinet wherein sufficient quantity of water was kept to maintain the temperature throughout the rearing period. The cabinet lid was kept partially open to allow free circulation of air inside the cabinet.

The results of observations on the development, growth of larvæ and pupæ and the fecundity of adults are summarised in Table I.

TABLE I

Effect of synthetic diet on the development of S. littoralis

Food	No. of larvæ in test	Percentage of survival	Mean developmental periods in days			Mean weight in mg.				Mean No. of eggs per female
			Larva	Pupa	Total	Full-grown larva	Pupa	Adult		
								Female	Male	
Castor leaf ..	50	30	14·44	13·00	27·44	880	278	303	199	381·2
Synthetic diet ..	50	92	14·59	12·13	26·72	920	518	483	227	452·6

The data show that though there is no great difference in larval period and pupal period of larvæ fed on the synthetic diet, synthetic diet has enhanced not only the weight of various stages of insects but also increased the fecundity of adults as compared to that of natural food. No difference in hatching of eggs of moths reared on natural food and moths reared on the synthetic diet was observed. The increased mortality in the insect reared on natural food is due to contamination by certain pathogens.

No fungal growth was observed on the diet kept continuously for 5 days. This may

be attributed to the addition of methyl-paraben.[4]

The acceptability of this diet to the castor semi-looper *Achæa janata* L. was also tested. It is interesting to note that the larvæ feed well on this diet.

The authors, are thankful to M/s. Sigma Chemical Company, U.S.A., for having provided some of the chemicals used in the present study.

Division of Entomology, V. T. SUNDARAMURTHY.
Agrl. College and T. R. SUBRAMANIAM.
Res. Inst., Coimbatore-3,
Tamil Nadu, *December* 26, 1968.

1. House, H. L., *Information Bull. No. 5*, Res. Inst., Canada, Dept. Agric., Onterio.
2. Nachaippan, RM., *M.Sc. (Ag.) Dissertation* submitted to the University of Madras, 1968.
3. Eldefrawi, M. E., Toppozada, A., Mansouy, N. and Leid, M., *J. econ. Ent.*, 1964, **57**, 591.
4. Colford, J. R., *Ibid.*, 1967, **60**, 1192.

LIGHT AND SPORULATION IN *CERCOSPORA PERSONATA* (BERK. *ET* CURT.) ELL. *ET* EVERH.*

Cercospora personata sporulates poorly in culture and loses sporulating character on repeated subculturing.[1] Selective subculturing from spores as well as special media have been recommended to secure sporulation in Cercosporæ. Leach[2] reported that light (visible or near ultraviolet) can induce sporulation in a variety of fungi. Recently, this was found to be effective in the case of *C. beticola*.[3] Two poorly sporulating isolates of *C. personata* made in 1959 and 1967 respectively by one of us (R. N. S.) and maintained on potato-sucrose agar were also induced to sporulate by light. No sporulation occurred if the cultures were incubated in total darkness but profuse sporulation was noticed when plates seeded with mycelial fragments from dark-grown cultures were kept under fluorescent lamps. Sporulation commenced even by the end of 48 hours if the mycelial bits used as inoculum were sufficiently large. If they were smaller, 3–4 days were required for sporulation to start. The medium used apparently had no influence on this since it occurred even on water agar.

It was also of interest to see whether the fungus needed light to sporulate on infected groundnut leaves. Leaves of groundnut variety TMV 2 showing lesions, about 4 mm. in diameter (25 days after inoculation), were washed in distilled water so as to remove any spores already present, blotted dry, and incubated in moist chambers in light and in darkness. After 48 hours the lesions were punched out carefully, suspended in a small volume of water and spores were brought into suspension by shaking with a few glass beads. Spore numbers were then determined with a hæmocytometer. The results are shown in Table I.

TABLE I

Effect of light on spore production by C. personata *on groundnut leaves*

Expt.	No. of spores/lesion	
	Light	Darkness
1	136,800 ± 7,100	68,200 ± 8,500
2	140,900 ± 4,000	63,600 ± 4,100

It is seen that light is not essential for sporulation on the host. However, it increases spore production. In *Helminthosporium gramineum* Houston and Oswald[4] have observed that light is not required for spore production on the host tissues though needed in pure culture. In the present experiment with infected leaves the light treatment was given only after formation of sporophores unlike in experiments on artificial media. This might be the reason why sporulation occurs even in darkness on leaves. This and other aspects of sporulation in *C. personata* are under investigation.

One of us (K. S. B.) thanks the University Grants Commission for the award of a Junior Research Fellowship.

University Botany Laboratory, K. S. BHAMA.
Madras-5, *August* 28, 1969. R. N. SWAMY.

* Memoir No. 72, from the Centre for Advanced Studies in Botany.

1. Shanta, P., *Proc. Indian Acad. Sci*, 1956, **44 B**, 271.
2. Leach, C. M., *Proc. Int. Seed Test. Ass.*, 1967, **32**, 565.
3. Calpouzos and Stallknecht, G. F., *Phytopathology*, 1967, **57**, 679.
4. Houston, B. S. and Oswald, J. W., *Ibid.*, 1946, **36**, 1049.

IDENTIFICATION OF SUBSTITUTED BENZOIC AND CINNAMIC ACIDS IN POLLEN GRAINS

THE accumulation of phenolic acids in various forms, free or as glycosides and esters, is known to occur in almost all plant tissues. These organic acids, which are generally synthesized[1,2] from aromatic amino-acid phenylalanine, have been regarded as metabolically inert substances because of their irreversible biosynthetic pathways. Hence, these acids behave as characteristic stable end products