ported agrees very well with what is known about the effect of auxin on root formation. The induction of roots on cuttings by basal application of indole acetic acid requires at least 100 times higher concentration than does apical application.

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THE GREEN MUSCARDINE FUNGUS ON THE PERIODICAL CICADA

IN connection with special work by the senior writer in the Department of Entomology, University of Maryland, on the periodical cicada, *Magicicada septendecim* (L.), it is of especial interest to note that the green muscardine fungus (*Metarrhizium anisopliae* (Metsch.) Sorokin) was found on this insect in the spring of 1936.

On May 22, 1936, Mrs. P. W. Wetmore, Takoma Park, Md., brought to the senior writer two live nymphs of the periodical cicada, both of which proved to be attacked by a fungus. The nymphs were sluggish and had failed to molt. Otherwise they appeared normal with no evidence of infection. When examined microscopically, however, abundant mycelium of a fungus with septate mycelium was found in them. Later, other similar specimens were collected on the ground from the same location. Four dead nymphs also were found in the tunnels. These latter were covered with fairly abundant creamy-white mycelium. This mycelium was transferred to potato-dextrose agar and to nutrient beef agar on which the fungus grew readily and subsequently sporulated abundantly. It also sporulated readily on rice kernels. The fungus on diseased nymphs placed in Petri dish moistchambers also sporulated abundantly in 5 to 19 days. The sporulating fungus was olive-green in color. From the microscopical characters of the spores and sporophores from these various sources, it was evident that the fungus was *Metarrhizium anisopliae* (Metsch.) Sorokin. The spores measured 1.8–4.5 × 7.8–12.8 μ, chiefly 3–3.8 × 9.7–11.3 μ. On the basis of these measurements, apparently the fungus is the long-spored form referred to by Delacroix,[1] Friederichs[2] and Johnston,[3] and named *f. major* by Johnston.

Apparently the short-spored form of *M. anisopliae* was found in Java on large singing cicadas by v. Höhnel,[4] who described the fungus as a distinct species, *Penicillium cicadinum*. Petch[5] transferred this species to the genus *Metarrhizium*, making the combination *M. cicadinum* (v. Höhnel) Petch. However, Petch, who also gives a literature summary, prefers to consider *M. cicadinum* as a synonym of *M. anisopliae*. This view seems tenable. The Java fungus, however, apparently represents the short-spored form of *M. anisopliae*, as its spores are described as 1.5–2 × 5–6 μ, rarely 7 μ.

Healthy nymphs and healthy adults were artificially inoculated with spores from the pure cultures of *M. anisopliae* kept in moist Erlenmeyer flasks, and both nymphs and adults became diseased, the nymphs being more susceptible than the adults. The fungus was readily reisolated. However, the fungus did not sporulate on the adults. Later, newly hatched nymphs also were inoculated in Petri dishes and these young nymphs proved to be unusually susceptible.

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THE EFFECT OF REPEATED CORTIN INJECTIONS UPON RENAL EXCRETION IN THE NORMAL ORGANISM[1]

IT has been reported[2] that large amounts of cortin produce a differential effect upon the excretion of electrolytes in normal human beings. A further study of these effects has been made in normal dogs.

Normal adult female dogs were fed a constant diet consisting of a mixture of beef heart, Purina chow and 2 g of NaCl daily. The dogs were fed at the same time every day, which was nine hours before the beginning of the test period. They were allowed as much water as they desired, except on the test days. During the tests the dogs were kept in metabolism cages and at three-hour intervals were catheterized and given 100

TABLE I

RENAL EXCRETION IN A DOG DURING THE FIRST SIX HOURS FOLLOWING THE INTRAVENOUS INJECTION OF CORTIN*

Date	Injection	*Vol.* cc	*Na* m.Eq.	*Cl* m.Eq.	*K* m.Eq.
1–21–37	Control	194	10.17	12.57	4.34
1–23–37	Control	184	6.18	9.39	3.95
1–25–37	Cortin	**130**	**1.24**	**4.83**	**6.25**
1–27–37	Cortin	**90**	**3.84**	**6.45**	**6.82**
1–29–37	Control	256	10.18	15.28	4.92
2– 3–37	Cortin	**132**	**5.67**	**9.87**	**7.22**
2– 5–37	Control	172	7.72	11.59	5.05
2–15–37	Cortin	**222**	**7.15**	**13.42**	**6.22**
2–26–37	Control	206	9.66	9.38	6.04
3– 4–37	Control	147	8.37	12.40	6.35
3– 9–37	Cortin	**243**	**13.46**	**18.52**	**6.07**

* Heavy-faced type, after cortin; light faced type, control. (20 cat units in 0.5 cc.)

[1] From the department of physiology, the Ohio State University, Columbus. Aided by a grant from the Rockefeller Foundation.

[2] G. W. Thorn, Helen R. Garbutt, F. A. Hitchcock and F. A. Hartman, *Proc. Exp. Biol. and Med.*, 35: 247, 1936.

[1] G. Delacroix, *Bull. Soc. Myc. France*, 9: 260–268, 1893.

[2] K. Friederichs, *Centbl. Bakt.* [etc.], Bd. 50, Abt. 2, (13/19): 335–356, 1920.

[3] J. R. Johnston, *Puerto Rico Bd. Commrs. Agr. Bul.* 10, 33 pp., 1915.

[4] F. v. Höhnel, *Sitzber. Akad. Wiss. Wien*, Math. Naturw. Kl. Bd. 118, Abt. 1: (1–178 in reprint), 1909.

[5] T. Petch, *Brit. Mycol. Soc. Trans.*, 16: 55–75, 1931.

cc of water by stomach tube. The maximum change in electrolyte excretion following cortin injection occurred during the first six hours. Initial injections of cortin (20 cat units) into eight normal dogs produced marked reduction in the sodium and chloride excretion and usually an increased excretion of potassium. The initial response of the different dogs to the same amount of cortin was essentially the same.

Four dogs have been injected repeatedly. With each succeeding injection the response has become smaller until it has entirely disappeared. Table I is typical of the four animals. Two human beings have shown a similar response to repeated cortin injections.

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SCIENTIFIC APPARATUS AND LABORATORY METHODS

A PLEURAL CANNULA[1]

THE recording of respiration in animal experiments is frequently a source of difficulty to the student and annoyance to the staff. The more accurate methods are beset with so many technical difficulties that either the student fails to obtain a record or the apparatus may be so expensive that the school fails to provide it. In an attempt to simplify our animal experimental methods, we have used many devices for graphically recording respiration. The pleural cannula method seemed to be the most simple, even though it involved a slight amount of surgical procedure, *i.e.*, a short slit in the skin between two ribs, a sharp thrust of the cannula so that the open end lies in the pleural cavity. The outer end of the cannula is connected with a recording tambour, giving essentially a record of variations in intrathoracic pressure during respiration. The most annoying difficulty with this method is the ease with which the cannula is displaced, thereby upsetting the record or requiring re-introduction when the cannula is completely pulled out. This unfortunate accident seems most likely to occur at the most critical moments of an experiment.

The principle of recording respiration by the pleural cannula method seemed sound and very easy to apply. The only difficulty is the instability of the cannula itself. A careless finger catching the rubber tube leading to the tambour resulted in the sudden withdrawal of the cannula; or even the accidental dropping of a light instrument on the rubber tubing may cause the cannula to slip out. It was felt that if we could overcome this difficulty of instability in this method, we would have an easily recording mechanism for respiration.

With this idea in mind, we developed the pleural cannula herein described. Instead of using the ordinary flattened and tapering point on the pleural cannula, we capped the point with a two-pointed barb which was somewhat flattened and pierced with four holes. Fig. 1 is a drawing of the cannula. One of the four holes is seen near the tip of the barb. This cannula is introduced in much the same manner as any pleural cannula, except that the flattened sides of the barb should be introduced parallel with the borders of the ribs. When the cannula enters the pleural cavity, it is immediately given a turn of 90°. This will result in the barb points catching the ribs if you attempt to withdraw it. While firmly holding the cannula in this position, quickly tighten the wing-nut shown at A in Fig. 1. If the wing-nut is very firmly tightened, the cannula will now stay firmly anchored in the pleural cavity and the rubber tubing may be quite carelessly handled without danger of the cannula slipping out. The firm tightening of the wing-nut on the underlying washer effectively seals and prevents loss of negative pressure in the pleural cavity.

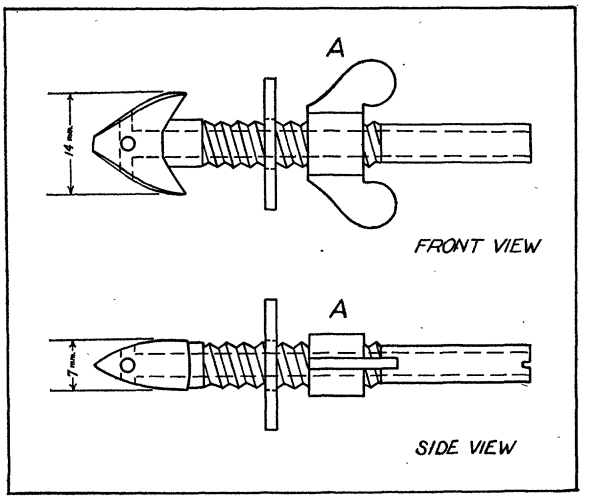

FIG. 1. Pleural Cannula

Certain precautions, if taken, will give better results. For example, it is best to insert the cannula where the rib borders lie in closest approximation; otherwise the barb will fail to engage the ribs and will come out. We usually choose the right side of the chest so that the heart will not be interfered with. The site chosen is about the midaxillary line and fairly high up on this line. It is best to cut the hair very short over the chosen site. It should be removed from a space about

[1] From the Department of Pharmacology of Boston University School of Medicine and the Evans Memorial Hospital.