Synthesis, Transport, and Utilization of Specific Flagellar Proteins during Flagellar Regeneration in *Chlamydomonas*

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**ABSTRACT** We labeled gametes of *Chlamydomonas* with 10-min pulses of $^{35}$S04$^{-2}$ before and at various times after deflagellation, and isolated whole cells and flagella immediately after the pulse. The labeled proteins were separated by one- or two-dimensional gel electrophoresis, and the amount of isotope incorporated into specific proteins was determined. Individual proteins were identified with particular structures by correlating missing axonemal polypeptides with ultrastructural defects in paralyzed mutants, or by polypeptide analysis of flagellar fractions. Synthesis of most flagellar proteins appeared to be coordinately induced after flagellar amputation. The rate of synthesis for most quantified proteins increased at least 4- to 10-fold after deflagellation. The kinetics of synthesis of proteins contained together within a structure (e.g., the radial spoke proteins [RSP]) were frequently similar; however, the kinetics of synthesis of proteins contained in different structures (e.g., RSP vs. $\alpha$- and $\beta$-tubulins) were different. Most newly synthesized flagellar proteins were rapidly transported into the flagellum with kinetics reflecting the rate of growth of the organelle; exceptions included a central tubule complex protein (CT1) and an actinlike component, both of which appeared to be supplied almost entirely from pre-existing, unlabeled pools.

Isotope dilution experiments showed that, for most quantified axonemal proteins, a minimum of 35--40% of the polypeptide chains used in assembling a new axoneme was synthesized during regeneration; these proteins appeared to have pre-deflagellation pools of approximately the same size relative to their stoichiometries in the axoneme. In contrast, CT1 and the actinlike protein had comparatively large pools.

When the flagella are amputated from many types of eucaryotic cells, the cells regenerate new flagella. This phenomenon has been frequently studied to learn more about the induction and regulation of synthesis of a defined set of proteins and how these proteins are transported and assembled into a developing cell organelle (12, 37, 39, 43, 44, 49). Several in vivo labeling studies have demonstrated that synthesis of the tubulins and other flagellar proteins is stimulated after flagellar loss (6, 15, 23). In vitro translation studies of isolated mRNA have shown that during regeneration there is an increase in the levels of translatable mRNAs for the tubulins and other flagellar proteins (24, 47), and studies using cDNA probes complementary to tubulin mRNA have provided evidence that tubulin mRNA is accumulated after flagellar amputation (30, 41). These findings are in good agreement with an in vivo study using an inhibitor of RNA synthesis which suggested that tubulin synthesis during ciliary regeneration requires new mRNA production (15). Formation of a new flagellum therefore appears to involve the transcription of specific mRNAs and their translation into proteins which are subsequently used to assemble the organelle or to replenish protein pools. Flagellar regeneration thus provides a model system for studying the regulation of gene expression as well as organelle morphogenesis and assembly.

Flagellar regeneration in the unicellular green alga *Chlamydomonas* is particularly suited to such studies. The flagella are easily detached from *Chlamydomonas*, and populations of cells will synchronously regenerate new flagella (39). The kinetics of regeneration have been well-characterized, and
inhibitor studies have clearly shown that normal regeneration is dependent on new protein synthesis (23, 39). The detached flagella can be readily isolated, free of cell body proteins and contaminating proteases (48, 51); this greatly facilitates analysis of the individual flagellar proteins. As a result, a large number of *Chlamydomonas* flagellar proteins have now been identified with either the flagellar membrane (19, 48) or with specific axonemal structures (11, 14, 18, 34–36, 48–50).

Although a considerable amount of information is available on the synthesis of the tubulins after flagellar loss in *Chlamydomonas* (7, 23, 24, 30, 41, 47), relatively little is known about the synthesis of the many proteins which are assembled into the flagellum in fixed ratios relative to the tubulins. In vivo studies have indicated that after deflagellation, the dyneins and several other axonemal proteins are synthesized with kinetics similar to that of the tubulins (23, 24); however, other flagellar proteins are synthesized with different kinetics (23). Analysis of in vitro translation products of mRNAs isolated from regenerating cells have shown that levels of translatable mRNAs for a large number of proteins are elevated in regenerating cells (24); several of these products comigrated with flagellar proteins, but they were not identified with specific flagellar structures.

To understand how the cell controls and coordinates the formation of a complex organelle such as the flagellum it is essential to know more about how the synthesis of the other axonemal components is regulated relative to that of the tubulins during flagellar regeneration. It is also important to have information on the relative pool sizes of the various flagellar proteins and their rates of transport into the growing flagellum. We have investigated these phenomena in *Chlamydomonas*. Our results, presented here, indicate that: (a) synthesis of most flagellar proteins is stimulated at the same time soon after deflagellation; (b) proteins contained within the same structure often appear to be coordinately synthesized, but (c) proteins in different structures are not coordinately synthesized; (d) most newly synthesized flagellar proteins are rapidly transported into the flagellum with kinetics which reflect the rate of growth of the organelle, and do not necessarily coincide with the time course of their synthesis in the cytoplasm; and (e) large pools of at least two flagellar proteins (the actinlike protein associated with the inner dynein arm, and a protein of the central tubule complex) are present in the cell body and are the source for most of the protein of these types used in assembling a new flagellum.

**MATERIALS AND METHODS**

**Materials**

$^{35}$SO$_4$ (as H$_2$SO$_4$, carrier free, 43 Ci/mg) was obtained from New England Nuclear (Boston, MA). DNase I and RNAse A were obtained from Boehringer-Mannheim Biochemicals (Indianapolis, IN).

*Dictyostelium* actin was kindly provided by Dr. J. S. Condeelis (Albert Einstein College of Medicine). Gel casting materials for electrophoresis were purchased from Bio-Rad Laboratories (Richmond, CA) unless noted otherwise. Carrier ampholytes (pH ranges 5–7 and 3–10) for isoelectric focusing were obtained from Bio-Rad Laboratories (Richmond, CA) at 16,000 rpm (31,000 g) for 20 min at 4°C; the pellet was prepared for electrophoretic analysis. The membrane and matrix proteins were recovered from the resulting supernatant fraction by acetone precipitation. The supernate was mixed with 9 vol of acetone prechilled in an EtOH-dry ice bath, the mixture was spun in the SS-34 rotor at 10,000 rpm (12,000 g) for 10 min at 4°C, and the supernate was removed. The residue, containing the membrane and matrix proteins, was prepared for electrophoresis.

**Whole Cell Solubilization**

Whole cells were prepared for electrophoretic analysis by the following procedure. The cells (6–10 × 10$^6$ cells) were pelleted and resuspended in 15 ml of a solution (modified from Howell [17]) containing 50 mM Tris HCl, pH 7.5, 200 mM NaCl, 1 mM EDTA, 2 mM MgCl$_2$, 80 mM β-mercaptoethanol, 1 mM PMSF (phenylmethanesulfonyl fluoride), and 1 mg/ml l-lysine. The cells were sedimented again and the pellet was frozen in an EtOH-dry ice bath for storage at −85°C until solubilization. The frozen cells were solubilized by a modification of the sonication procedure of O'Farrell (32). The major changes were the addition of 2 mM PMSF and 1 mg/ml l-lysine to the sonication buffer, the addition of 0.25% NP-40 during the nuclease digestion, and a low-speed centrifugation (IEC 253 rotor [Damon/IEC Division, Needham Heights, MA], 2000 rpm, 1,100 g, for 10 min at 4°C) to remove any nonsolubilized material.

**Flagellar Amputation and Regeneration**

Gametes were placed in a Virtis 100-ml fluted glass flask and deflagellated by shearing for 75 s in a Virtis Model 23 homogenizer set at full speed. The cells were then placed on a magnetic stirrer under a fluorescent lamp and allowed to regenerate. For determination of regeneration kinetics, aliquots of cells were removed at 5-min intervals and fixed by mixing 1:1 with a solution of 2.5% glutaraldehyde in isotonic saline diisent (Fischer Scientific Co., Fair Lawn, NJ). Flagellar lengths on a minimum of 25 cells were then measured using an ocular micrometer on a Zeiss Universal microscope outfitted with Nomarski interference contrast optics at ×800.

**Isotopic Labeling of Chlamydomonas**

Vegetative cells were labeled by growing them for 4–5 days to a density of 3–4 × 10$^9$ cells/ml in MM modified to contain 4.8 × 10$^{-2}$ mM SO$_4$$^{2-}$ and 1.0 mM H$_2$SO$_4$/125 ml. Steady-state labeled gametes were obtained by transferring labeled vegetative cells into 125 ml of low SO$_4$$^{2-}$/NFM containing 43 μCi H$_2$SO$_4$ and allowing gametic differentiation to proceed. Under these conditions, the specific activity of $^{35}$SO$_4$$^{2-}$ was the same in both MM and low SO$_4$$^{2-}$/NFM. These procedures resulted in a specific activity of between 1000–2000 cpm/μg of TB-precipitate protein. Gametes were pulse labeled by placing 5-ml aliquots of a gametic cell suspension in 25-ml Erlenmeyer flasks containing $^{35}$SO$_4$$^{2-}$ (0.5 mCi carrier-free H$_2$SO$_4$ per 5 ml of medium) plus 25 μg/ml chloramphenicol; after 10 min, the pulse was stopped by addition of unlabeled MgSO$_4$ to a final concentration of 3.2 mM. Close adherence to our growth and labeling conditions was necessary to achieve uniform rates of uptake of label at the various times throughout regeneration.
Measurement of Incorporation into TCA-Precipitable Protein

The incorporation of isotope into TCA-precipitable protein was measured by a batch filter paper disc method (Rosenbaum et al. [39], as modified from Mans and Novelli [28]). The background was <0.3% of the maximum number of counts among the filters.

Protein Determination

The protein concentration in various samples was determined by the method of Bradford (5), using the microassay described in the Bio-Rad Protein Assay Dye Reagent Kit (Bio-Rad Laboratories), and with bovine gamma globulin as the protein standard.

Gel Electrophoresis

One-dimensional electrophoresis of proteins in SDS polyacrylamide gels was carried out by the method of Laemmli (20, 21), modified to give better resolution of the high molecular weight components of the whole flagella. The plug consisted of 8% acrylamide; the separating gel (32 cm long by 16 cm wide by 1.5 mm) was 3-15% linear acrylamide gradient, 0-8% linear glycerol gradient; and the stacking gel (2 cm long) was 3% acrylamide. The running buffer contained 0.024 M Tris base, 0.192 M glycine, and 0.13% SDS. Samples were prepared by boiling for 1 min in a sample buffer containing 0.124 M Tris HCl, pH 6.8, 10% glycerol, 2% SDS, 0.001% Bromophenol blue, and 1% β-mercaptoethanol. The samples were electrophoresed at 60 V until the tracking dye was 1 cm into the separating gel, at which time the voltage was increased to 120 V until the dye front reached the plug (~16 h).

The flagellar components were also analyzed by two-dimensional gel electrophoresis (NEPHGE SDS). The methods used were those of O'Farrell et al. (33), except that the second dimension used the SDS polyacrylamide gel system described above.

All gels were initially fixed in 20% methanol (MeOH)-7.5% acetic acid (HAc). If the gels were to be stained, they were then washed with 35% ethanol-7.5% HAc for 24 h to remove the ampholytes, and subsequently washed with 20% MeOH-7.5% HAc followed by staining for 3 h in 45% MeOH and 9.0% HAc containing 0.25% (wt/vol) Coomassie Brilliant Blue. After destaining in 20% MeOH-7.5% HAc, the gels were photographed using Kodak High Contrast Copy or Panatomic X films. For the quantification procedure described below, the destained gels were washed with 2.5% HAc just before excision of relevant spots.

Fluorography

One- and two-dimensional gels containing 35S-labeled proteins were prepared for fluorography according to the procedure of Bonner and Laskey (4) and Laskey and Mills (22).

Quantification of Two-dimensional Gels

To determine the amount of 35S in specific flagellar components, axoneme, whole flagella, or whole cell samples were electrophoresed with unlabeled axonemal protein (75 µg/gel unless stated otherwise) as a carrier. After staining, spots corresponding to particular proteins were excised, solubilized with NCS Tissue Solubilizer (Amersham/Searle Corp., Arlington Heights, IL) (1), and counted. Control experiments using two-dimensional gels in which various amounts of labeled material were loaded along with the carrier showed that the recovery was linearly proportional to the counts loaded (see Results).

RESULTS

Identification of Structural Proteins in the Flagellum

The Chlamydomonas flagellum is composed of a membrane-bounded axoneme containing nine outer doublet microtubules, two central microtubules, and a number of microtubule-associated structures, including the inner and outer dynein arms, the radial spokes and spoke heads, and the projections of the central tubules (Fig. 1). When analyzed by two-dimensional gel electrophoresis, the flagellum is seen to contain α- and β-tubulin and over one hundred other proteins (Fig. 2a). To determine which of these proteins are associated with particular axonemal structures, we have (a) compared the proteins of isolated axonemes of wild-type with those of paralyzed mutants containing known ultrastructural defects in order to correlate missing proteins with missing structures in the mutants and (b) analyzed the polypeptides of various flagellar fractions isolated using selective solubilization procedures.

Fig. 2b shows a fluorograph of a two-dimensional gel of axonemes obtained by treating isolated flagella with the detergent NP-40 to dissolve the flagellar membrane; note that several proteins are present in greatly reduced amounts relative to whole flagella (Fig. 2a). These proteins (arrows in Fig. 2a) represent components of the flagellar membrane and matrix. Two of these proteins, having molecular weights of 65,000 and ~125,000, (m and m' in Fig. 2a), have been identified as major membrane components by analysis of membrane vesicles isolated using the procedure of Witman et al. (48) (results not shown; see reference 38).

To identify proteins associated with the central tubules, the central tubuleless mutants pf-18 and pf-19 were analyzed. In these mutants, the central tubules are replaced by a core which runs longitudinally down the center of the axoneme (46); this core is frequently lost when the axonemes are isolated (48, 50, 51). Fig. 3 shows fluorographs of two-dimensional gels of axonemes of wild-type and mutant pf-19. A number of proteins were found to be reproducibly missing in both the pf-18 and pf-19 axonemes; five of these are indicated in Fig. 3. These proteins presumably represent components of the central tubule–central sheath complex. The four high molecular weight (HMW) proteins previously shown by Witman et al. (51) to be associated with the central tubule complex were not resolved in the two-dimensional gels.

Similarly, to identify components of the radial spokes, mutants pf-14, missing the entire radial spoke (50, 51), and pf-1, missing only the spoke head (34), were analyzed. 14 proteins...
FIGURE 2 Fluorographs of two-dimensional gels of whole flagella (WF) (a), and axonemes (AX) (b) labeled to steady state with 

\[ ^{35}S \] NE, NEPHGE dimension, SDS, SDS PAGE dimension. Very little of the 65,000 and 125,000-mol wt membrane proteins (a: m and

\[ m' \], respectively), remain in the axoneme preparation b. Arrows in a indicate minor membrane and matrix proteins. The major high

molecular weight membrane protein (48) is not resolved in our two-dimensional gel system. Gels were loaded with ~200,000 cpm

and exposed for 2.5 d. Inset in b shows a shorter exposure of the tubulin region. Identification of the upper and lower tubulin

spots as \( \beta \)- and \( \alpha \)-tubulin, respectively, is based on their isoelectric points and the relative amounts of \( ^{35}S \) taken up into their

cysteine and methionine under steady state conditions (42, 45, and see text). \( \beta \)-tubulin may run ahead of or behind \( \alpha \)-tubulin in

SDS acrylamide gels, depending on the source of SDS used (2; and see Fig. 13). The molecular weight standards (\( \times 10^3 \)) used for

calibration of the gels were cytochrome C (11,700), ovalbumin (43,000), bovine serum albumin (68,000), phosphorylase A (94,000),

and myosin (220,000).

(R1-R14, Fig. 4a) were found to be reproducibly missing in pf-14 axonemes (not shown). These proteins are probably all components of the radial spokes. Six of the 14 proteins missing in pf-14 (proteins R1, R2, R7, R8, R10, and R11) were also missing in pf-1 (not shown), indicating that these proteins are contained in the spokehead. These results are in good agreement with previous studies of the protein deficiencies in pf-14 and pf-1 axonemes (34, 51).

Proteins associated with a unique group of three protofila-

ments which form the wall between the lumens of the A and B tubules of the outer doublets (48) were identified by analysis of the isolated partitions on two-dimensional gels (results not shown) (13; C. R. Gattass and G. B. Witman, manuscript in preparation). Four of these proteins relevant to this study are indicated in Fig. 4a.

A number of low and medium molecular weight proteins

associated with the 12s and 18s dynein ATPases were identified

bcoelectrophoresing the purified dyneins (11) with isolated

axonemes. Four of these components analyzed in the present

study are indicated in Fig. 4a. One protein (A, Fig. 4a), possibly associated with the 12s I dynein contained in the inner

arm (10), ccoelectrophoresed with actin isolated from Dictyostel-

ium (38). This agrees with reports that an actinlike component

can be isolated from axonemes (35), and that a protein with an
electrophoretic mobility identical to that of actin is missing in

mutant pf-23, which lacks the inner arm (18).

Fig. 4 summarizes the proteins of interest to this study which

have been identified with particular structures in the axoneme.

With the exception of the \( \alpha \)- and \( \beta \)-tubulins, each of these proteins comprises a small percentage of the axonemal protein
FIGURE 3  Fluorographs of NEPHGE-SDS gels of axonemes of wild-type (1132D) and the central-tubuleless mutant pf-19. The numbered arrows in the wild-type gels indicate some of the proteins missing in the mutant; the positions of these deficiencies are indicated by numbered arrows in the mutant fluorograph. ~200,000 cpm were loaded and the gels exposed for 2.5 d. Inset: Portion of a fluorograph of another gel of wild-type axonemes. Polypeptides CT1, CT2, and CT5 are more clearly resolved from the tubulins (α, β) at this lower loading and shorter exposure. 100,000 cpm, 1.5-d exposure.

in either Coomassie Blue-stained gels or fluorographs of 35S-labeled proteins. Fig. 4b shows the results for an 35S-labeled preparation run on a two-dimensional gel and quantitated as described below. The identified minor proteins individually contained 0.03% to 0.26% of the TCA-precipitable radioactivity loaded onto the gels. These values do not necessarily represent the relative amounts of these proteins in the axoneme, but reflect (a) the relative amounts of sulfur-containing amino acids in the proteins and (b) the relative amounts of the proteins entering our gel system (some material does not enter the first-dimensional gel).

Experimental Design

Gametic cells rather than vegetative cells were used for studies on protein synthesis during flagellar regeneration for several reasons. Gametic cells appear to have smaller pools of unassembled flagellar protein than vegetative cells. Cycloheximide-treated gametes regenerate flagella of only one-fifth to one-tenth the normal length (23), as compared with the one-half normal length flagella of vegetative cells so treated. Gametes also have a 10-fold higher rate of $^{36}\text{SO}_4^{2-}$ incorporation into TCA-precipitable protein (unpublished result). Both factors result in an increase in the specific activity of proteins labeled during regeneration. Moreover, deflagellated gametic cells regenerate flagella more slowly than vegetative cells (23). The time required for regeneration is 140 min for gametes as compared to 90 min for vegetative cells. The final length of the regenerated flagella is approximately the same in both gametes and vegetative cells. This longer regeneration time makes possible increased temporal resolution in pulse label experiments.

The kinetics of flagellar regeneration in a population of gametes is illustrated in Fig. 5. The time periods chosen for pulse labeling are superimposed on the curve. These periods included a time before deflagellation (to determine basal levels of synthesis, transport, and assembly of flagellar proteins), several times during the regeneration process, and a time when the flagellum has nearly regained its normal steady-state length. The rate of incorporation of $^{36}\text{SO}_4^{2-}$ into TCA-precipitable protein before deflagellation and at the different times during regeneration are shown in Fig. 6; the rate varied by no more than ±12% within an experiment. After the pulse, whole cells, whole flagella, or flagellar fractions were isolated and electrophoresed on one- and two-dimensional gels.

For quantification, these labeled samples were run together with unlabeled axonemal carrier protein on two-dimensional gels. Fig. 7 shows a Coomassie-Blue-stained gel of an 35S-labeled whole cell preparation coelectrophoresed with unlabeled axonemal material. It should be pointed out that although some of the heavily labeled spots in the fluorographs were not always well resolved due to β-particle spreading and saturation of the emulsion, the flagellar components appeared as discrete spots in the stained gels (cf. Figs. 7 and 10 a, 30-min gel). Spots of interest were excised, solubilized, and counted, all as described in the experimental procedures.

To check the accuracy of our quantitation procedures over a range of cpm/spot, we determined the amount of label in the α- and β-tubulins on gels loaded with different amounts of 35S-labeled axonemes together with the standard amount of unlabeled axonemal carrier protein (75 μg). The radioactivity recovered was linearly proportional to the radioactivity loaded for both species of tubulin (Fig. 8). Similar results were obtained for other proteins quantitated. The difference in incorporated counts between the α- and β-tubulins is due to the
FIGURE 4 (A) Fluorograph of NEPHGE-SDS gel of wild-type axonemes summarizing proteins relevant to our study which were identified by analysis of mutants and flagellar fractions. (B) Quantification of the label in components of axonemes labeled to steady-state with 35S and coelectrophoresed with 50 μg of unlabeled axonemes. Percentages were calculated relative to the total TCA-precipitable material loaded per gel and do not reflect the relative amounts of each protein in the axoneme (see text). α, β: α- and β-tubulins, respectively; R1-R14: radial-spoke proteins; A: actinlike component; CT1-CT5: central-tubule proteins; P1-P4: partition components; 12s, 18s: proteins associated with the 12s and 18s dyneins,

difference in cystine and methionine content between the two proteins (42, 45).

One cannot assume that the same proportion of each flagellar protein enters the gel system. However, because the values obtained for individual proteins are directly proportional to the amount of radioactive protein loaded, comparison of the

FIGURE 5 Regeneration kinetics for a population of gametes deflagellated at time zero, with the periods (labeled PD and 1-7) of pulse labeling superimposed on the regeneration curve. Initial length of flagella was 12 μm.

FIGURE 6 Incorporation of 35S04-2 into whole gametes before deflagellation and during regeneration. Counts per minute incorporated into TCA-precipitable protein (per 25-μl aliquot of the cell suspension) is plotted against time into the pulse for each of the pulse periods (1-7 and PD) indicated in Fig. 5. The pulses were done in the presence of chloramphenicol to block chloroplast and mitochondrial protein synthesis. Chloramphenicol had no effect on the kinetics of regeneration, and a comparison of the patterns of synthesis of several proteins during regeneration in the presence and absence of chloramphenicol indicated that the drug did not affect the synthesis of flagellar proteins. The calculated linear regression line (△) is also shown.

respectively; m: residual amount of the 65,000-mol wt membrane protein.
Relative amounts of radioisotope in spots at different times during regeneration will give an accurate indication of the relative timing and amounts of increases or decreases of label within or between specific proteins.

**Synthesis of Flagellar Proteins During Regeneration**

To study the time of induction of flagellar proteins and their rates of synthesis at various times after deflagellation, regenerating gametes were pulse labeled at the previously indicated times and whole cells isolated and analyzed by gel electrophoresis. Fig. 9 shows a fluorograph of a one-dimensional gel of these samples along with a whole flagellar standard. Uptake of label into the α- and β-tubulins was substantial as early as 15 min after deflagellation and reached a peak 30–50 min into regeneration (see below for quantitation). The 65,000-mol wt membrane protein was labeled most heavily at the earliest time point. Label appeared in the high molecular weight dynein components only during the second and subsequent time points. Some minor proteins (e.g., see arrow in Fig. 9, right hand margin) were maximally labeled near the end of regeneration.

To achieve better resolution of individual proteins, these samples were analyzed on two-dimensional gels. Portions of the fluorographs are shown in Fig. 10a. As noted earlier, the overall rate of incorporation of $^{35}$S remained constant during regeneration; the increase in the amount of isotope in the region of the gels shown was offset by a complementary decrease in other portions of the gels (see Fig. 9).

The first panel in Fig. 10a (PD) shows protein synthesis in the nondeflagellated cells. Substantial synthesis occurred for the α- and β-tubulins, the central tubule complex protein 1 (CT1), and the 65,000-mol wt membrane protein. This synthesis was not due to flagellar regeneration in a subpopulation of the cells which might have spontaneously lost their flagella; light microscope examination of >100 cells showed that before deflagellation all cells had fully grown flagella. Nor was the synthesis due to slight flagellar growth in the entire population of cells; gametic flagella undergo virtually no change in length over several hours under our experimental conditions (38).

The synthesis of most flagellar components was coordinately stimulated following flagellar amputation. The gel patterns for the regenerating cells showed that tubulin synthesis was greatly elevated for the first 50 min after deflagellation, and then steadily declined to predflagellation levels. The central tubule proteins CT1 and CT2 were not well resolved (in the fluorographs) during the early times, but CT2 synthesis clearly peaked between 15 and 50 min, and then declined to background levels by 90 min into regeneration (circle, 90 min). Synthesis of the 65,000-mol wt membrane protein (m) and the radial spoke proteins (marked by arrows in 15-, 30-, and 50-min gels) also peaked early during regeneration. A large number of nonflagellar proteins of molecular weight <65,000 were induced after deflagellation; these may represent "stress-in-
duced proteins” similar to those synthesized in starved *Tetra
hymena* as a response to deciliation, heat shock, or release
from anoxia (16). In *Chlamydomonas*, several proteins are
induced by heat shock; the same proteins are a subset of those
induced after flagellar amputation (29).

The changes in the uptake of label into certain of the flagellar
components were quantitated as described above. Fig. 10 b and
c show the results for the α- and β-tubulins, the 65,000-mol wt
membrane protein, the radial spoke proteins 1–4, the central
tubule complex proteins 1 and 2, and the actinlike component.

Synthesis of the tubulins peaked between 30 and 50 min into
regeneration (Fig. 10 b: α, β); the rate of tubulin synthesis at
this time was approximately 10-fold higher than the predefla-
gellation rate. The relatively high level of incorporation of
isotope into α-tubulin compared to β-tubulin may reflect pref-
erential synthesis of the α-tubulin (cf. Figs. 4 and 8).

The 65,000-mol wt membrane protein reached a maximum
rate of slightly more than fourfold its basal level of synthesis at
~30 min into regeneration. It subsequently showed a more
gradual decline in labeling than did the tubulins. The rates of
synthesis of radial spoke proteins 1–4 peaked by 15 min and
decayed by 30 min. Because the amount of label recovered
from the radial-spoke proteins at the other times approached
background levels, the rate of synthesis of these proteins before
deflagellation and late in regeneration could not be accurately
estimated. However, the data do indicate that after deflagella-
tion their rate of synthesis rapidly increased to at least 10-fold
the predeflagellation rate, and then rapidly decreased to rela-
tively low levels.

Incorporation of label into the central tubule protein CT2
increased fourfold to a peak by 15 min and then dropped
rapidly, approaching predeflagellation levels by 90 min into
regeneration. Uptake of label into CT1 increased fivefold to a
maximum by 30 min and gradually declined.

In contrast, the actinlike protein showed slightly more than
a twofold increase in labeling after deflagellation. This was by
far the smallest increase observed for any flagellar protein. The
possibility exists that other actin species are contained in this
spot and contribute to the basal level of synthesis; one species
might then be elevated several-fold during regeneration, but
the increase in the spot as a whole would appear much smaller.
In any case, other studies (see below) indicate that nondefla-
gellated cells contain a relatively large pool of actin available
for flagellar formation, and that new synthesis makes a negli-
gible contribution to the actin used in assembly of a new
flagellum.

This analysis of protein synthesis in whole cells indicated
that at least several flagellar proteins, including α- and β-
tubulin, the 65,000-mol wt membrane protein, and CT1, were
continuously synthesized at low levels in nonregenerating ga-
etes. After deflagellation, the synthesis of the majority of
flagellar proteins was stimulated at least four- to tenfold; for
most proteins, this enhancement began at the same time (within
the limits of our time resolution, which was ~15 min). Proteins
contained within any one type of axonemal structure (such as
the radial spokes) usually had similar kinetics of synthesis, but

![Figure 9](image-url) Fluorograph of a 5–15% acrylamide gradient gel of whole cell proteins from regenerating cells prepared at the indicated times during regeneration. Cells were pulsed with 35SO₄²⁻ for 10 min immediately before their isolation. Each channel was loaded with an approximately equal number of cells. PD: predeflagellation sample; WF: steady-state-labeled whole flagella; α, β: α-, and β-tubulin, respectively; m: 65,000-mol wt membrane protein; D: high molecular weight dynein components; arrow (right hand margin) indicates a component labeled late in regeneration. 55,000 cpm/channel; 2-d exposure. Molecular weight standards (X 10³) were the same as for Fig. 2.

![Figure 10](image-url) (a) Portions of fluorographs of two-dimensional gels of whole cell proteins processed before deflagellation (PD) and at the indicated times during regeneration. Cells were pulsed with 35SO₄²⁻ for 10 min immediately before isolation. ~1.25 x 10⁶ cells containing 10⁶ cpm were loaded per gel; 24-h exposure. α, β: α-, and β-tubulin, respectively; m: 65,000-mol wt membrane protein; CT1, CT2: proteins of the central tubule–central sheath complex; arrows 1–4: (15- to 50-min gels); radial-spoke proteins 1–4. The circle in the 90-min gel indicates the position of CT2, which is not visible beyond 70 min. Because a large number of cpm
were loaded on the gels (10^6 cpm vs. 5.5 x 10^6 cpm in Fig. 9) to permit detection of minor components, some regions of the fluorographs are not well resolved. This loss of resolution was due to saturation of the emulsion and the resulting coalescence of spots in the fluorographs, and was not a problem inherent in the gels themselves; at shorter exposures the major components were well resolved. This smearing thus did not affect our ability to quantitate the gels. (b and c) Quantification of label in flagellar proteins in whole-cell samples from predelflagellation and regenerating cells. Data is shown for the α- and β-tubulins (α, β), the 65,000-mol wt membrane protein (m), the radial-spoke proteins (R1–R4), central-tubule proteins 1 and 2 (CT1, CT2), and the actinlike protein (A) probably associated with the 12s dynein ATPase. (b) also shows the means (X, X') and standard deviations (I) for the pooled data from this and another experiment where the α- and β-tubulins were quantitated.
groups of proteins contained within different structures were often synthesized with dissimilar kinetics.

Transport of Newly Synthesized Proteins into the Flagellum

The above experiments established the timing and rates of synthesis of a number of flagellar proteins. To determine the timing and rates of transport of these newly synthesized proteins into the flagellum, cells were pulsed at various times prior to and during regeneration and the flagella were isolated and prepared for electrophoretic analysis.

Fig. 11 shows a fluorograph of a one-dimensional gel of these samples. Label appeared in the α- and β-tubulins at the earliest time period and peaked by 30 min into regeneration. Most of the proteins followed the tubulin pattern. However, in a number of proteins the appearance of isotope either: (a) was delayed relative to the tubulins (e.g., proteins 1 and 2 [the high molecular weight membrane component], and the high molecular weight dynein components) and declined more slowly (protein 1); (b) peaked earlier (proteins 3, 4, and 5); or (c) peaked later (proteins 6 and 7) during regeneration. Label was taken up into the 65,000-mol wt membrane protein in much greater amounts than would be expected from the relative amount of label incorporated into it under steady-state conditions; this may indicate that the cytoplasmic pool for this component is relatively small compared to those for most other proteins. Note that the high molecular weight membrane protein (protein 2) shows a later uptake of label during regeneration than does the 65,000-mol wt membrane component.

These flagellar samples were also analyzed in two-dimensional gels. Equal numbers of flagella were run on the gels; Fig. 12 shows portions of the resulting fluorographs. Label appeared in most of the previously identified proteins at one time or another during regeneration. Duplicate gels were quantitated as previously described (Fig. 12 b–d).

Nonregenerating Flagella: The first panel of Fig. 12 a shows the transport of labeled proteins into nonregenerating flagella. Label appeared in the 65,000- and 125,000-mol wt membrane proteins and in components which comigrated with the α- and β-tubulins. As previously mentioned, this uptake was not due to formation of new flagella in a subpopulation of the cells nor to continued elongation of previously formed flagella. The appearance of label in proteins under these conditions must therefore represent turnover of these proteins in the flagellum. Further evidence for this comes from the fact that the spot pattern in the tubulin region is different in nonregenerating and regenerating flagella (see below).

To determine whether this turnover was confined entirely to the membrane and matrix, or whether some turnover occurred in the axoneme, nonregenerating cells were pulse labeled for
FIGURE 12 (a) Portions of fluorographs of two-dimensional gels of whole flagella isolated before deflagellation (PD) and at the indicated times during regeneration. Equal numbers of flagella (~2 x 10⁷) were loaded on each gel; the total cpm loaded are shown in the lower right corner of each panel. The gels were exposed for 2 d. a, ß: a- and ß-tubulin, respectively; m and m’, 65,000 and 125,000-mol wt membrane components, respectively; CT2–CT4: central-tubule-complex proteins; 1–14: radial-spoke proteins; P1–P4: proteins associated with the partition protofilaments; 12s, 18s: 12s I and 18s dynein components, respectively; B–E: unidentified proteins discussed in the text; Ac, CT7: positions where the actinlike component and CT7 were expected; X: axonemal component with mobility similar to ß-tubulin in gels run with Bio-Rad SDS. (b–d) Quantification of the appearance of label into specific proteins within the flagellum during regeneration. a, ß: a- and ß-tubulin, respectively; m: 65,000-mol wt membrane component; R1–R4: radial-spoke proteins; CT1, CT2, CT5: central tubule complex proteins; A: the actinlike component; x____x, the average rate of flagellar growth (μm/min) during each of the pulse periods.
Portions of fluorographs of two-dimensional gels prepared using SDS from Bio-Rad Laboratories (panels a, c, e, g, and i) or the Baker Chemical Co. (panels b, d, f, h, and j). T. WF, T. AX, and T. M + MX: whole flagella, axoneme, and membrane and matrix fractions respectively. All were isolated from nondeflagellated cells immediately after a 10 min pulse with $^{35}$S-O$_4$. AX, steady-state $^{35}$S-labeled axonemes; Reg. WF, whole flagella isolated from regenerating cells immediately after an $^{35}$S-O$_4$ pulse from 5–15 min after flagellar amputation (similar to 15 min time point, Figure 12 a). $\alpha$, $\beta$, $\alpha$- and $\beta$-tubulins, respectively; m and m', 65,000 and 125,000 mol wt membrane proteins, respectively; X, component with a mobility similar to $\beta$-tubulin in the presence of Bio-Rad SDS. Material recovered from approximately equal numbers of flagella were loaded on gels corresponding to panels c–h; exposures in cpm are shown in the lower right hand corner of each panel.

Comparison of the spot patterns obtained for the whole flagella sample in the presence of the two brands of SDS (Fig. 13 c and d) showed that, although $\alpha$- and $\beta$-tubulins were present in the preparation, the majority of the label comigrating with $\beta$-tubulin in the presence of Bio-Rad SDS did not shift mobility in the presence of Baker SDS and therefore was not $\beta$-tubulin but rather some other component (component "X"). An even greater proportion of the label incorporated into the axonemes was in this component and did not shift mobility in the presence of Baker SDS (cf. Fig. 13 e and f); only a very small amount of label was present in $\alpha$- and $\beta$-tubulin, and this label was evenly distributed between the two tubulins. In the membrane and matrix fraction, some label was contained in component X, but much more was present in $\alpha$- and $\beta$-tubulin (cf. Fig. 13 g and h). Component X was not observed in steady-state-labeled axonemes (Fig. 13 a and b), indicating that it is present in the axoneme in very small amounts relative to the tubulins. Furthermore, it was not observed in regenerating whole flagella isolated from cells pulsed from 5–15 min (Fig. 13 i and j) or from 20–30 min (not shown) after deflagellation, indicating that the majority of the label comigrating...
with α- and β-tubulin in regenerating flagella (see below) is in authentic α- and β-tubulin.

**REGENERATING FLAGELLA.** In the regenerating flagellum, uptake of labeled α- and β-tubulin and the 65,000-mol wt membrane protein peaked at 30 min (Fig. 12a and b). The relative amounts of newly synthesized 65,000-mol wt membrane component and α- and β-tubulins transported into the flagellum did not reflect the relative rates of synthesis for these components (compare Figs. 10b and 12b); as noted previously, the comparatively high rate of transport for the 65,000-mol wt membrane component suggests that there is a smaller usable pool for this protein. The uptake of labeled 65,000-mol wt membrane protein decayed at a slightly slower rate than that of the tubulins; these kinetics are probably due to the fact that synthesis of the membrane component did not drop off as rapidly during regeneration as did that of the tubulins (see Fig. 10b). Although the maximum rate of tubulin synthesis occurred at 30–50 min, the maximum rate of transport of labeled tubulin into the regenerating flagellum occurred at 15–30 min, apparently reflecting the rate of growth of the organelle, which was greatest during the pulse period ending at 30 min (Fig. 12b).

All of the components of the partition protofilaments were most heavily labeled at 30 min (Fig. 12a). However, there were differences in the kinetics of appearance of label into these components: between 15 and 30 min, there was a more rapid increase in the amount of label in P1 and P4 than in P2 and P3 (quantification not shown). These results suggest that the partition components are either noncoordinately synthesized early in regeneration or that there are considerable differences in their relative pool sizes.

Most of the radial-spoke proteins showed a maximum uptake of label at approximately 30 min into regeneration. The relative amounts of label in components R1–R3 were similar to the amounts present under steady-state-labeling conditions (cf. Fig. 4; R4 > R2 ≥ R3 > R1); however, component R4 contained comparatively low levels of label (Fig. 12c). This reflects the low rate of synthesis (Fig. 10b) and relatively large pool for component R4 (see below).

Uptake of label into central tubule proteins CT2, CT3, CT4, and CT5 also was greatest at ~30 min (Fig. 12a and d). In contrast, CT1 showed a nearly constant low level of labeling throughout regeneration.

Several components of the 12s I and 18s dyneins showed a maximum uptake of label at 30 min into regeneration (Fig. 12a). However, at no time during regeneration was significant label detected in the actinlike protein associated with the 12s I dynein.

The very low level of isotope in CT1 and the absence of isotope in the actinlike component of regenerating flagella was not due to lack of synthesis of either of these proteins (Fig. 10c), nor to insufficient amounts of newly synthesized 12s I dynein or central tubule material being transported into the flagellum (Fig. 12a, 30- and 50-min gels). Moreover, both the actinlike component and CT1 were observed in Coomassie
Blue stained gels of regenerating axonemes (38; results not shown), indicating that both components were transported into the flagellum in an unlabeled form during regeneration. These results suggest that there is a relatively large pre-existing pool for each of these components in nondeflagellated cells (see below).

Spots B–E (Fig. 12a) represent unidentified proteins which were synthesized and transported into the flagellum during regeneration, but were not observed in fluorographs of steady-state labeled whole flagella or in Coomassie Blue stained flagellar preparations. Pulse-chase studies in which cells were pulsed from 15–25 min after deflagellation and their newly formed flagella isolated at 25, 140, and 200 min showed that components C and D were labeled at 25 min and underwent neither an increase nor decrease in labeling at the later times, even though the amount of isotope in total flagellar protein and most individual flagellar components increased fivefold between 25 min and completion of regeneration at 140 min (38; results not shown). Proteins C and D therefore appear to be stable components of the flagellum, but are synthesized and transported into the organelle early in regeneration and in such small amounts that they are not readily detected in steady-state-labeled flagella. By 140 min, the flagella had reached their steady-state length and the pattern of labeling had returned to that seen before deflagellation.

These studies indicated that most newly synthesized flagellar proteins are transported into the regenerating flagellum with kinetics which reflect the rate of growth of the organelle, and that the kinetics of appearance of a labeled protein in the flagellum does not necessarily coincide with the time course of its synthesis in the cytoplasm. Exceptions included CT1 and the actinlike protein, which appeared to be transported into the flagellum from pre-existing, unlabeled pools.

**Pool Sizes for Specific Axonemal Proteins**

The results of our studies on the transport of newly synthesized proteins into the regenerating flagellum suggested that for certain axonemal proteins there were substantial differences in the relative sizes of their pools in the cell body. To obtain more information on the sizes of the preflagellation pools of these proteins, isotope dilution experiments were carried out in which vegetative cells were grown in low SO$_4^{2-}$ minimal medium in the presence of $^{38}$SO$_4^{2-}$, and transferred to low SO$_4^{2-}$ nitrogen free medium (NFM) containing the same specific activity of $^{38}$SO$_4^{2-}$ (see Materials and Methods). The next day the resulting gametes were placed in NFM containing the normal concentration of unlabeled SO$_4^{2-}$. The cells were then immediately deflagellated and allowed to regenerate flagella for 140 min. A portion of these cells were processed to give isolated axonemes (1st Reg.); the remainder were deflagellated a second time, allowed to regenerate for 140 min, and the newly regenerated axonemes then isolated (2nd Reg.). As controls, axonemes were isolated from a portion of the initial nondeflagellated population of gametes (ND + 0 min), and from a nondeflagellated population after a 3 h chase (ND + 180 min).

With each deflagellation and regeneration, the specific activity (cpm/µg TCA precipitable protein) of the isolated axonemes decreased by an amount equal to 35–40% of the specific activity of the controls (dashed line in Fig. 15a and b). If there were no cytoplasmic pools for any of the flagellar proteins, this decrease would indicate that the chase was 35–40% effective during the first regeneration and 70–80% effective during the second regeneration. However, gametic cells are known to contain cytoplasmic pools sufficient to form at least 1–2 µm of flagella in the absence of new protein synthesis (23); these values represent the pool size for the limiting protein, so the total amount of unassembled flagellar protein in the cytoplasm could be considerably greater. Therefore, the chase was probably even more effective than the percentages given above. For example, the decrease in specific activity of the axonemes in this experiment could have resulted from a chase which was 80% efficient, if the average pool size was equivalent to two three-quarter length flagella and 80% of this pool was used during each regeneration.

Equal numbers of isolated axonemes were then loaded on two-dimensional gels. Fig. 14 shows portions of the resulting fluorographs; Fig. 15 shows the quantitation for some of the proteins.

The controls (Fig. 14, ND + 0 min and ND + 180 min) indicated that most of the axonemal proteins are stable components which do not undergo much turnover during a 3-h period. Any proteins undergoing a rapid turnover would have shown a loss of label in the chased controls.

In axonemes regenerated after the first deflagellation (Fig. 14, 1st Reg.), the amount of label in most of the components was visibly less than in the controls. This difference was even greater in axonemes regenerated after the second deflagellation (Fig. 14, cf. ND + 180 min. and 2nd Reg.). Quantification of similar gels indicated that, with each deflagellation and regeneration, the amount of label in the $\alpha$- and $\beta$-tubulins and radial spoke proteins 1–3 decreased 35–40% relative to the control values (Fig. 15a and b); the sizes of the pools of these proteins (relative to their stoichiometries in the axoneme) must therefore be approximately the same. Such a decrease in the $\alpha$- and $\beta$-tubulins was expected in as much as these proteins make up a
substantial proportion of the total axonemal protein. In contrast, relatively high levels of label persisted in radial spoke protein 4, central-tubule protein 1, and the actinlike protein, even after two deflagellations and regenerations (Fig. 15a and b). This indicates that comparatively large pools of these proteins must be present in nondeflagellated gametes, and is in good agreement with the results of our studies on the transport of these proteins into the flagellum.

DISCUSSION
Paralyzed mutant and flagellar fractionation studies have allowed a large number of Chlamydomonas flagellar proteins to be identified with specific structures. This report has examined the synthesis and transport of some of these proteins in nondisassembled and regenerating cells to determine whether the synthesis of flagellar proteins is coordinately stimulated and regulated after amputation, and to determine the extent to which new protein synthesis and pre-existing protein pools are involved in flagellar regeneration.

Several proteins, including α- and β-tubulin and the 65,000-mol wt membrane protein, were found to be synthesized at low levels in nonregenerating gametes; these same proteins were also the major species transported to the flagellum of nonregenerating cells. Flagellar fractionation studies showed that most of these labeled proteins were contained in the membrane and matrix fraction. Turnover of components found in this fraction probably represents replacement of proteins lost by blebbing of membrane vesicles from the flagellar tip (3). There are two possible explanations for the presence of α- and β-tubulins in this class of proteins: (a) tubulin may be directly associated with the Chlamydomonas flagellar membrane, as has been reported for ciliary membranes of Tetrahymena (9) and Aequipectin (44), or (b) unassembled tubulin may be present in the flagellar matrix and become trapped within the membrane vesicles as they are blebbed from the flagellum; the tubulin lost might then be replaced by newly synthesized tubulin. With respect to the latter possibility, it should be noted that Levy (25) has proposed a model in which flagellar growth control is related to the continuous loss of components from the flagellar tip; as the flagellum elongates, this loss, coupled with the increasingly longer time required for diffusion of components down the flagellum, would result in slowing and eventually cessation of growth as the concentration of an essential component decreased and finally dropped below a critical value at the growing tip.

The only protein which turned over rapidly in the axoneme was component X, a protein which comigrated with β-tubulin in the presence of Bio-Rad SDS but was resolved from the tubulins in gels containing Baker SDS. This was a very minor component which could not be detected in steady-state-labeled axonemes and consequently would not have been noticed in our isotope dilution experiments. It would be of considerable interest to know more about this component and its role in the flagellum. Lefebvre et al. (23) have provided evidence that Chlamydomonas can "sense" the shortening or absence of its flagella and responds by initiating flagellar protein synthesis; component X could be involved in this "signaling" process if flagellar loss results in its rapid accumulation in the cell body.

In deflagellated cells, synthesis of most flagellar proteins was stimulated at least four- to tenfold above predeflagellation levels; in the case of some of the minor components, such as the radial-spoke proteins, it could not be determined whether this increase represented an induction from a true "off" state or an elevation from basal levels so low they could not be detected in our experiments. In any case, for most proteins this stimulation appeared to be initiated coordinately (within the limits of our time resolution) during the first 15 min after deflagellation. This is in accord with the observation of Lefebvre et al. (23) that a number of components comigrating with unidentified flagellar proteins were induced within 30 min after deflagellation. However, we also observed a few flagellar proteins which were not synthesized until late in regeneration (see Fig. 9).

Synthesis of the α- and β-tubulins increased rapidly after deflagellation and peaked between 30 and 50 min at a rate at least 10-fold the predeflagellation rate. This 10-fold increase in the level of tubulin synthesis is in good agreement with in vitro studies which have shown that the amount of tubulin mRNA available for in vitro translation is ~10-fold higher in regenerating Chlamydomonas than in nonregenerating cells (41). Moreover, the kinetics of tubulin synthesis after flagellar amputation closely parallels changes in the levels of mRNA coding for tubulin; hybridization studies using cloned tubulin probes have revealed that the amount of tubulin mRNA in total or polyadenylated RNA begins to increase 5–8 min after deflagellation, and peaks after 30–50 min at levels ~25–40-fold that found in nondisassembled cells (30, 41). The close correspondence between the timing of tubulin synthesis and tubulin mRNA production strongly suggests that tubulin synthesis is controlled at the transcriptional level. That the increase in tubulin mRNA appears to be several fold greater than the increase in tubulin synthesis may indicate that at least tubulin mRNA is translated at a lower efficiency during flagellar regeneration. A general decrease in translational efficiency, such as might occur due to crowding of the translational machinery by a large number of newly synthesized mRNAs coding for flagellar proteins, could account for the observed reduction in synthesis of many nonflagellar proteins after deflagellation (23; see Fig. 9). Confirmation of this will require further study to resolve the apparent discrepancy between the measured values for translatable vs. total tubulin mRNA in cells before and after deflagellation.

At first examination, the induction pattern for most other flagellar proteins appeared to follow that of the tubulins, but quantification of these patterns revealed significant differences. For example, synthesis of the radial-spoke proteins peaked at about 15 min after deflagellation (vs. 30–50 min for the tubulins), and synthesis of central tubule protein 1 increased only fivefold above predeflagellation levels (vs. a 10-fold increase for the tubulins) and subsequently did not drop off as rapidly as did that of the tubulins. These findings indicated that production of proteins contained in different parts of the flagellum or axoneme is not coordinately regulated. However, the kinetics of synthesis of certain proteins contained within an axonemal structure (e.g., the α- and β-tubulins, or the radial-spoke proteins R1–R4) were very similar, suggesting that coordinate regulation may occur for certain proteins which are topographically very closely related.

The fact that the synthesis of most flagellar proteins is not coordinate beyond the initial induction implies that different mechanisms must be involved in regulating the production of the tubulins and at least some of the minor components—or groups of minor components. This conclusion is supported by the observation that the compound APM (amiprophos methyl) selectively inhibits tubulin synthesis in regenerating Chlamydomonas but appears to have no effect on the production of several other flagellar proteins (7). Whether the apparent dif-
shock proteins must therefore be quite complex, rather than a maximum rates of synthesis at different times and declining at induction of all heat-shock polypeptides begins within 10 min in D. melanogaster, heat shock markedly stimulates the post-translational phenomenon such as rapid degradation of 630 proteins. In D. melanogaster, heat shockmarkedly stimulates the distribution along the length of the flagellum. Alternatively, such the flagellum only during a slightly later period in regeneration, Fig. 12, which were synthesized and rapidly transported into central microtubule cap (8). Similarly, components C and D of the flagellum in significant amounts in a labeled state. This was not due to a lack of transport of these components into the regenerating flagellum, as both were clearly visible in Coomassie-Blue-stained gels of axonemes isolated from the regenerating organelle. Neither is it likely that a delay in transport, such as might have been caused by posttranslational processing, prevented the newly synthesized polypeptides from reaching the flagellum during the relatively short period of the pulse. In studies in which cells were labeled from 15-25 min after deflagellation (the peak periods of synthesis for these components) and then chased for 115 and 175 min before isolation of the flagella, still no label was detected in these spots (38). The results strongly suggested that large pools of these components are present in the cell body and supply most of the actin and CTI used during flagellar assembly.

This conclusion was confirmed by our isotope dilution studies which indicated that nondeflagellated gametes contain relatively large cytoplasmic pools of central tubule protein 1 and the actinlike component. Nondeflagellated cells also appeared to contain a large pool of radial-spoke component 4, a finding in good agreement with our data on the rates at which this component is synthesized and transported into the flagellum. All other components quantitated, including α- and β-tubulin, radial spoke components R1-R3, and central-tubule protein 2, had smaller cytoplasmic pools which were all approximately the same size relative to their stoichiometries in the axoneme. For these latter proteins, a minimum of 35-40% of the polypeptide chains used in constructing a new axoneme were newly synthesized; an even larger percentage was used if the chase in these experiments was <100% efficient.

A somewhat different result was obtained by Stephens (44) in studies on the synthesis of axonemal proteins and their transport into growing cilia of sea urchin embryos. Deciliation of Strongylocentrotus droebachiensis embryos stimulates the synthesis of many axonemal proteins; however, the majority of these proteins are not used during the subsequent formation of a new axoneme, but rather go to replenish cytoplasmic pools depleted during the regeneration event. These pools appear to be quite large; Stephens (44) estimated that the pools for most minor components and for the tubulins of the B-tubule are 5-10 times larger than needed to form a new set of cilia. In contrast, we found that in Chlamydomonas gametes a large amount of the protein used to construct a new axoneme is synthesized during regeneration. Moreover, for most proteins a maximum of only 10-30% of the polypeptide chains used in assembling a new axoneme after two deflagellations were present in the cytoplasm before the first deflagellation, suggesting that existing pools are largely depleted by formation of one set of new flagella. Therefore, compared to sea urchin embryos, Chlamydomonas maintains small pools of flagellar proteins and relies quite heavily on protein synthesis for the formation of new flagella. These differences are probably due to the particular requirements of each organism. As Stephens hypothesized (44), it may be energy efficient for a sea urchin embryo to synthesize and store ciliary proteins prior to cilogenesis for use not only in the initial formation of cilia by the
blastomereres, but also in later elongation of apical cilia and for replacement of lost cilia. However, the flagella of Chlamydomonas do not continue to elongate after their initial period of growth, and loss of the flagella in gametes of Chlamydomonas is probably a rare event in nature. It would, therefore, be most efficient for Chlamydomonas to maintain minimal cytoplasmic pools of flagellar proteins and synthesize a substantial amount of these proteins de novo should production of new flagella become necessary.

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REFERENCES

1. Abrue, S. L., and R. L. Brünter. 1975. Synthesis of tubulin and actin during the development of the mouse. Exp. Cell Res. 114:115-141.

2. Baxandall, J., R. A. Fornen, and T. Birthing. 1979. Electrophoretic patterns with β-tubulin trailing in SDS-free polyacrylamide gels. J. Cell Biol. 83(1, Pt. 2):2339-2341 (Abstr.).

3. Bergman, K., U. W. Goodenough, D. A. Goodenough, J. Jawitz, and H. Martin. 1975. Genetic differentiation in Chlamydomonas reinhardii. II. Flagellar membranes and the aggregation reaction. J. Cell Biol. 76:606-622.

4. Bonner, M., and R. A. Laskey. 1974. A film detection method for trinitium-labeled proteins and nucleic acids in polyacrylamide gels. Eur. J. Biochem. 46:83-88.

5. Bradford, M. 1976. A rapid and sensitive method for the quantitation of microgram quantities of protein utilizing the principle of protein dye binding. Anal. Biochem. 72:248-254.

6. Brown, D. L., and K. Rogers. 1978. Hydrostatic pressure-induced internalization of flagellar axonemes, disassembly, and reuse during flagellar regeneration in Polyommia. Exp. Cell Res. 117:313-324.

7. Collis, P. S., and D. P. Weeks. 1978. Selective inhibition of tubulin synthesis by amino- prophyll ethoxybenzyl during flagellar regeneration in Chlamydomonas reinhardii. Science (Wash. D.C.) 202:440-444.

8. Dentler, W. L., and J. L. Rosenbaum. 1977. Flagellar elongation and shortening in Chlamydomonas. II. Structures attached to the tip of flagellar microtubules and their relationship to the directionality of flagellar microtubule assembly. J. Cell Biol. 74:747-759.

9. Dentler, W. L., M. M. Pratt, and R. E. Stephens. 1980. Microtubule-membrane interactions in cilia. II. Photosensitive cross-linking of bridge structures and the identification of a membrane-associated dynein-like ATPase. J. Cell Biol. 84:381-403.

10. Fay, R. B. 1980. The flagellar ATPases of Chlamydomonas reinhardii. Ph.D. Thesis. Princeton University.

11. Fay, R. B., and G. B. Witman. 1977. The localization of flagellar ATPases in Chlamydomonas reinhardii. J. Cell Biol. 75(2, Pt. 2):2326-2328 (Abstr.).

12. Fütter, H., and J. Kowit. 1977. Programmed synthesis of flagellar proteins during cell division in Chlamydomonas. J. Mol. Biol. 11:2407-2419.

13. Gattass, C. R., and G. B. Witman. 1979. Protein composition of the partition protofilaments of Chlamydomonas reinhardii. J. Cell Biol. 76:729-747.

14. Gattas, C. R., and G. B. Witman. 1979. Protein composition of the partition protofilaments of Chlamydomonas reinhardii. J. Cell Biol. 76:729-747.

15. Gattas, S. H., P. S. Collis, E. E. Young, and D. P. Weeks. 1981. Tubulin induction in C. reinhardii. J. Biol. Chem. 254:3084-3090.

16. Gattas, S. H., P. S. Collis, E. E. Young, and D. P. Weeks. 1981. Tubulin induction in C. reinhardii. J. Biol. Chem. 254:3084-3090.

17. Gattas, S. H., P. S. Collis, E. E. Young, and D. P. Weeks. 1981. Tubulin induction in C. reinhardii. J. Biol. Chem. 254:3084-3090.

18. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas. J. Cell Biol. 85:258-272.

19. Huang, B., U. K. Laemmli, and M. F. Harfe. 1975. Mutations of the head of bacteriophage T4. DNA packaging events. J. Mol. Biol. 85:375-385.

20. Huang, B., U. K., and M. F. Harfe. 1975. Mutations of the head of bacteriophage T4. DNA packaging events. J. Mol. Biol. 85:375-385.

21. Huang, B., U. K., and M. F. Harfe. 1975. Mutations of the head of bacteriophage T4. DNA packaging events. J. Mol. Biol. 85:375-385.

22. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

23. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

24. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

25. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

26. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

27. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

28. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

29. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

30. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

31. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

32. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

33. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

34. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

35. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

36. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.

37. Huang, B., G. Piperno, and D. J. L. Luck. 1979. Paralyzed flagellar mutants of Chlamydomonas reinhardii defective for axonemal microtubule amts. J. Mol. Biol. 254:3091-3099.