Polarized Insertion of New Membrane from a Cytoplasmic Reservoir during Cleavage of the Drosophila Embryo

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Abstract. Cellularization of the Drosophila embryo is a specialized form of cytokinesis that results in the formation of a polarized epithelium. The mechanisms of membrane growth during cytokinesis are largely unknown. It is also unclear whether membrane growth and polarization represent distinct processes that occur simultaneously or whether growth of the membrane is involved in the emergence of polarity. Using a combination of surface labeling and particles tracking techniques, we monitored the dynamics of marked membrane regions during cellularization. We find that the major source of membrane is intracellular, rather than in the form of a plasma membrane reservoir. Depolymerization of microtubules inhibits the export of a newly synthesized transmembrane protein from the Golgi apparatus to the plasma membrane and simultaneously blocks membrane growth. Membrane insertion occurs in a defined sequence at specific sites, first apical, then apical–lateral. Diffusion of the membrane appears insufficient to compete with the massive local insertion of new membrane. We thus identify a tightly regulated scheme of polarized membrane insertion during cellularization. We propose that such a mechanism could participate in the progressive emergence of apical–basal polarity.

Key words: cytokinesis • polarity • membrane growth • epithelial cells • cellularization

Introduction

An important challenge in cell biology is to understand the mechanisms underlying the establishment and maintenance of cell polarity. Many different cell types show a polarized organization reflected in the asymmetric distribution of proteins and lipids in the plasma membrane and the asymmetric organization of the microtubule and actin cytoskeleton. This organization is key to their functional specialization. For example, it is essential during polar bud growth in the yeast Saccharomyces cerevisiae, polarized growth during axon formation, and the transport of nutrients across endothelial barriers. Some cases involve the imposition of polarity on previously existing membranes whereas others are associated with new membrane growth. In spite of obvious differences in the process, many components of the polarization pathway are shared across species and the mechanisms have been the subject of intense cell biological and biochemical investigations (for a review see Drubin and Nelson, 1996). However, it remains a challenge to integrate the available information into a dynamic picture where the behavior of organelles and the cytoskeleton is monitored as polarity arises and where the hierarchy of decisions is reconstructed through the discovery of key regulators.

Cellularization of the Drosophila blastoderm is a remarkable process where the formation of a polarized epithelium is coupled to the process of cytokinesis. It offers an opportunity to understand the mechanisms of membrane growth during furrowing. It also allows one to ask whether membrane growth can contribute to the emergence of membrane polarity or whether these represent distinct processes. The first 13 nuclear divisions of the Drosophila embryo occur in a syncytium, resulting in 6,000 peripheral nuclei located beneath the plasma membrane. During cellularization, the membrane surface increases ~25-fold, invaginates between the nuclei, and ultimately yields 6,000 epithelial cells 30 µm tall. Most studies so far have focussed on the cytoskeletal rearrangements that control cellularization (Schejter et al., 1992; Foe et al., 1993). A systematic search for genes zygotically required for cellularization (Merrill et al., 1988; Wieschaus and Sweeton, 1988) led to the discovery of three essential loci that control the integrity of the actin cytoskeleton at the invagination front called the furrow canal (FC); serendipity-alpha (sry-a) (Schweisguth et al., 1990), nullo (Rose and Wieschaus, 1992), and bottleneck (bnk) (Schejter and...
Rned by membrane growth under situations of rapid cytokinesis (for a review see Glotzer, 1997). An alternative model proposes that the polarized distribution of microtubules might exert a force pushing the membrane and forcing it inward (Foe et al., 1993). These models constitute the current framework for our understanding of cellularization. However, neither model addresses the origin of the membrane required to accommodate the massive increase in membrane surface during cellularization. An attractive idea proposes that much, if not all of the membrane, exists as a reservoir in the form of villous projections of the plasma membrane (Fullilove and Jacobson, 1971; Turner and Mahowald, 1976). A alternatively, membrane growth could derive from the secretory pathway (Loncar and Singer, 1995). In both models the membrane has hitherto only been viewed as a passive aspect of furrowing, whether it is pulled downward by the actomyosin network or pushed inward by microtubules.

At the onset of gastrulation, the resultant epidermal cells have the hallmarks of polarized cells with adherens junctions separating the apical and basal-lateral domains. It is still unknown how this polarity is established. It may occur during cleavage of the embryo when the membrane grows and invaginates between the nuclei or, following the classical example of epithelial Madin-Darby canine kidney (MDCK) cells, it could occur after cellularization when contacts between cells trigger the formation of adherens and septate junctions. Nonpolarized MDCK cells use E-cadherin-based cell-cell contacts as an external cue to position and to trigger the assembly of adherens junctions (Drubin and Nelson, 1996). A dherens junctions in turn act as a fence to isolate two membrane domains that are subsequently the site of specific membrane targeting. Proteins travelling along the biosynthetic pathway are sorted at the level of the trans-Golgi network and directed to either the apical or the basal-lateral membranes. In addition, the endosomal compartment receives and redirects lipids and proteins to their apical or basal-lateral destinations. Thus, the adherens junctions serve both as a trigger of polarization and a device to maintain polarity. In neurons, the axonal and somatodendritic membranes are thought to be the analogue of the apical and basal-lateral membranes on the basis of similar protein targeting profiles (Dotti and Simons, 1990). Vional cytoplasmic flow occurs at sites of future membrane growth and precedes axon formation (Bradke and Dotti, 1997). Despite these similarities, neurons are devoid of adherens junctions and must resort to different strategies to maintain the integrity of adjacent membrane domains. Intramembrane diffusion barriers exist and involve interactions between the membrane and the underlying actin and spectrin cytoskeleton (Winckler et al., 1999). The most obvious difference between neurons and MDCK cells is membrane growth, which is extensive in the former and minimal in the latter.

The work presented here uses Drosophila to address whether the polarization of epithelial cells can be governed by membrane growth under situations of rapid membrane mobilization. In other words, is polarity reflected in the regulated insertion of proteins and associated lipids at different locations as the membrane grows, or does polarity appear subsequently when nonpolarized cells interact in a manner akin to MDCK cells? To address these issues, we developed techniques that allow the visualization of the flux of membrane proteins in living embryos during cellularization. We compared these dynamic patterns to the distribution of a newly synthesized transmembrane protein in stage-fixed embryos. We conclude that growth of the plasma membrane stems from the re-mobilization of ER- and/or Golgi-derived membrane populations that insert at precise locations in a regulated manner. The localized membrane delivery we identify in our experiments is such that lectin-labeled membrane patches and microbeads bound to an intact membrane are consistently displaced away from the sites of insertion. This suggests a mechanism in which polarization of the plasma membrane might be inherently linked to the polarized pattern of membrane growth during embryonic cleavage.

### Materials and Methods

#### Fly Strains

Oregon R stocks were used as wild-type controls in all of our experiments. To generate embryos zygotically deficient for Neur from mothers with a wild-type Neur dosage, we crossed C(3)se females with males heterozygous for Df(3L)81k19.

#### Surface Label of Living Embryos with Wheat Germ Agglutinin

Embryos were collected at 25°C on agar plates after 30-min collections. E embryos were aged 90 min at 25°C, dechorionated in 30% bleach for 2 min, rinsed with water, and lined up on a strip of agar. E embryos were then transferred on a coverslip covered with a thin layer of glue. After dehydration, the embryos were covered with 5700 halocarbon oil where they continued to develop normally.

At the desired stage, embryos were injected with lectin as follows. Wheat germ agglutinin (WGA) coupled to the Alexa488 fluorochrome (Molecular Probes, Inc.) was kept as a 1 mg/ml stock solution in PBS at −20°C. WGA was thawed, diluted 1:3–1:5 in PBS, and centrifuged at 14,000 rpm for 10 min. It was loaded in a fine capillary needle and injection was conducted in the perivitelline space at 50% egg length (further information available upon request from the authors at tiecult@molbio.princeton.edu). The coverslip was mounted on an inverted Zeiss LSM 510 confocal microscope where embryonic development proceeds normally. A II images were processed using A dobe Photoshop® software.

#### Coating, Injection, and Tracking Fluorescent Microspheres in Living Embryos

0.5-μm YellowGreen fluorescent carboxylated microspheres (Polysciences, Inc.) were coated with WGA-Alexa488 using the carbodiimide kit for covalent coupling (Polysciences, Inc.). WGA coated beads were diluted in PBS and injected in the vitelline space and followed under the confocal microscope. YellowGreen fluorescent beads were detected using a relatively large pinhole to be reliably tracked in 2-μm optical slices. The maximum area of contact between the beads and the membrane was calculated to be 0.25 × 0.25 × 3.14 = 0.19 μm². The membrane surface at the onset of phase 3 was largely underestimated, with the approximation to a smooth cylinder 6.5 μm in diameter and 5 μm high: 2 × 3.25 × 3.14 × 5 = 102 μm². This gave a minimum estimate of the relative surface of ~1/537.

#### Antibody Staining

A ntidye staining against N eurotactin (N eur), A maduillo (A rm), and myosin required heat fixation, which was described in Peifer et al. (1994). For embryos preinjected with colcemid, most of the halocarbon oil was removed manually; what remained was desolved in heptane. The heptane...
was replaced with a boiling NaCl buffer (68 mM NaCl, 0.03% Triton X-100) for 5–10 s. Injected embryos cannot be devitellinized with methanol/heptane and the vitelline membrane was removed instead with a sharp tungsten needle. Other fixations for antibody staining to Discs lost (Dlg), Discs-large (Dlg), and β-tubulin were done using standard protocols with 15% formaldehyde in PBS/heptane for 15 min, followed by methanol popping except for injected embryos.

In all cases, antibody staining was performed in BBT (PBS/0.1% BSA/0.1% Tween 20), except for an initial blocking step with 10% BSA in PBS and 0.1% Tween 20. Primary antibodies were incubated overnight at 4°C with the following dilutions: rabbit anti-Dil 1:1000 (B hat et al., 1999); rabbit anti-DIg 1:200; rabbit anti-I-Myo 1:4000 (generous gift of C. Fields); monoclonal mouse anti-NEP (BP106) 1:5 (Hybridoana Bank); monoclonal mouse anti-A rm 1:50; rabbit anti-A rm 1:200 (Pfeifer et al., 1994); mouse anti-β-tubulin 1:500 (Sigma Aldrich), rabbit anti-β-tubulin 1:1200 (Rippe et al., 1994). Subsequent reactions were performed at room temperature. Preadsorbed secondary antibodies coupled to A lex488, A lex546 (Molecular Probes, Inc.), and Cy3 were used at 1:500. The DNA dye H oeichst was used at 0.1 μg/ml for 5 min at the end of the staining protocols. Embryos were mounted in A qua Poly M ount (Polysciences, Inc.).

Colcemid Injections

Dechorionated embryos were lined up and dehydrated as described above. Colcemid (Sigma-A iRich) was stored as a 5 mM stock solution at 4°, diluted in water to 1.25 mM. Injection with an Eppendorf transjector allowed a reproducible result with an estimated final concentration ~12.25 μM around the middle of the embryo. The effect was uniform around the circumference of the embryo at the injection site. Lower concentrations of colcemid showed variable effects and microtubules were not depolymerized reproducibly (data not shown). Injection was performed at 50% egg length at 16–18°C.

Results

Four Phases in the Formation of a Polarized Epithelium

Cleavage of the syncytial blastoderm begins on entry into interphase of the 14th nuclear cycle and lasts ~70 min at 20°C. Living embryos can be followed with differential interference contrast (D I C) microscopy as shown in Fig. 1. Immediately after mitosis 13 nuclei, reappear as spheres 5 μm in diameter beneath the plasma membrane (Fig. 1 A, arrow). The cell surface above each nucleus protrudes in a dome shape known as a somatic bud, a feature of the membrane during interphase 9–14 (Foe and Alberts, 1983). During the cellularization process that follows, four phases can be observed, each characterized by a distinct feature of membrane invagination. Phase 1 lasts 10 min and results in the formation of the FC, a structure corresponding to the tip of the invaginating membrane. The nuclei also start to elongate basally. In the next three phases, each 20 min long, the FC moves inward at an increasing rate. In phase 2, the speed is very slow and the cellularization front remains in an almost steady position 5 μm basal to the surface of the embryo (Fig. 1, B and C, arrow). During this phase, the nuclei complete their elongation. In phase 3, invagination becomes more obvious, although still at a relatively slow rate until it reaches the basal part of the nuclei (Fig. 1 D, arrow). The invagination rate then abruptly increases by a factor of two in phase 4 and in the next 20 min gives rise to cells 35–50 μm tall (Fig. 1 E). Previous subdivision of the process into slow and fast phases corresponds to phases 2/3 and 4, respectively (Foe and Alberts, 1983).

The initial stages of cellularization can be followed with greater accuracy using a membrane-attached PDZ-con-
herence of the embryo, and remains unbound in the vitelline space (data not shown). We then used this labeling technique to characterize membrane synthesis, the rationale being that if new intracellular membrane is inserted, it will be seen as unlabeled membrane that either dilutes or displaces "old" labeled membrane. We first injected WGA–Alexa488 during phase 1. Initially, WGA is detected over the entire surface of the somatic buds (Fig. 2A, top), reflecting the numerous villous projections observed in electron microscopy (Fullilove and Jacobson, 1971; Turner and Mahowald, 1976). By the end of phase 1, WGA is no longer detected in the regions above the nuclei (Fig. 2A, dashed line), suggesting that this membrane is rapidly renewed by incorporation of unlabeled membrane from the interior. WGA label is still visible between the nuclei where it forms interconnected rings whose positions correspond to the future site of membrane invagination. This label coincides with the FC (Fig. 2C, arrowheads) and remains associated with the cellularization front as it moves inward (Fig. 2B). The basal displacement of this staining during cellularization confirms the subdivision of cellularization into four distinct phases based on the rate of membrane invagination (Fig. 1). Several time points are shown in different colors in Fig. 2B. In phase 2 (+12' and +14'), the label barely moves within 2 min. In phase 3 however, it moves inward of ~1 µm within 2 min (+33' and +35'). The label is still detectable late in phase 4 and moves much faster (+56' and +58').

We conclude from these observations that two adjacent membrane regions form in phase 1 that show distinct behaviors (Fig. 2D): the surface above the nuclei shows a rapid flux or turnover, whereas the FC defines a more stable membrane region that persists throughout the course of cellularization. Our data do not distinguish whether the persistent high levels of staining in the FC in phase 1 reflect only low turnover rates in that structure compared with the apical membrane or the active relocalization of the label from the apical membrane to the FCs. With the caveat that WGA may alter the normal behavior of the plasma membrane, these data suggest that the FC forms as a separate membrane region. Consistent with this view, note that Dlt becomes restricted to and remains in the FC throughout cellularization (Fig. 1) and that a basal junction with high levels of Arm forms at the boundary between the FC and the adjacent membrane of the somatic bud (Fig. 1). This junction may be required to isolate the FC from the rest of the plasma membrane.

**Apical Insertion of New Membrane and Lateral Transfer of Old Membrane**

The FC can actually only be labeled during phase 1.
WGA–Alexa488 injected during phase 2 appears to be ex-
cluded from the canal (Fig. 3, B and B′, arrowhead), although
it binds uniformly to the remainder of the embryo’s sur-
face (Fig. 3A, purple). A’s membrane invaginates at a slow
speed during phase 3, the label is gradually depleted from
the apical membrane while the lateral label persists and
the size of the labeled domain even expands (Fig. 3A,
blue; compare double arrow at time 0 and 118).

Figure 2. Confocal sections through
living embryos after injection with
WGA–Alexa488. All pictures show the
embryo from the side. N, nuclei. The
embryos were grown at 25°C, a temper-
ature that increases the rate of invagi-
nation compared with 20°C as shown in
Fig 1. (A and B) WGA label of the
same embryo at different time points
shown in different colors. At time 0
(A), WGA is broadly distributed at the
plasma membrane surface. 14 min
later, WGA is only detected between
nuclei, and the apical membrane
(dashed line) is unlabeled. This stain-
ing persists throughout invagination
(B) and is shown 2 min apart in phase 2
(+12′ and +14′), phase 3 (+33′ and
+35′) and phase 4 (+56′ and +58′).
(C) Another embryo where WGA (ar-
rowhead, top) colocalizes with the FC
(bottom, arrowhead). (D) Scheme il-
ustrating the changing distribution of
the initial label (red) from its apical dis-
tribution to the FC. The green mem-
brane represents the newly inserted un-
labeled membrane. Bar, 5 μm.

The edge of the lateral labeled patches maintain their
association with the invaginating FC, and the labeled
regions behave as though they simply accompany the
inward growth of the membrane until the end of cellular-
ization. In fact, the original most intensely labeled mem-
brane regions move basally at the same speed as the FCs
through phases 3 and 4. This suggests that there is no ob-
vious incorporation of unlabeled membrane at the invagi-
nation front or that if it occurred it would not contribute
to the growth of the plasma membrane. Instead, new
membrane incorporation appears to occur predominantly
apically and accounts for the bulk of the membrane
growth during phase 3.

**Lateral Insertion of New Membrane Becomes
Predominant in Phase 4**

The rate of membrane invagination greatly increases in
phase 4. We find that the pattern of membrane insertion is
very different from that characterized for the preceding
stages. When WGA label is injected at the end of phase 3,
fluorescence is initially detected both apically and laterally
(Fig. 3C, red and middle at time 0). In contrast to the
earlier stages, the apical membrane maintains its label,
though intensity slightly decreases, as cellularization pro-
ceeds. A gap of unlabeled membrane emerges in the apical
portion of the lateral region (arrowhead in Fig. 3C, 12 min
after time 0). This gap grows in size and the lateral mem-
brane previously labeled at time 0 is displaced basally and
maintains its association with the FC (not shown). We con-
clude that the point of new unlabeled membrane insertion
changes during phase 4, shifting from predominantly api-
cal to predominantly lateral. Given the mild reduction of
the apical membrane label, we conclude that apical inser-
tion still persists, but our data show that the major site of
insertion is lateral.

The changes in distribution of the labeled lectin at the
membrane surface do not reflect a change in distribution
of the WGA receptors under either normal or experimen-
tal conditions. For instance, if lateral transfer of WGA
during phase 3 stemmed from increasing levels of WGA
receptors laterally as a result of local changes in mem-
brane composition, injection of WGA at the end of phase
3 would predominantly label the lateral membrane. How-
ever, our data show that the ability of WGA to bind the
apical membrane appears constant at any time during cel-
larization (compare Fig. 3, A and C, at time 0). Clear-
ance of fluorescent WGA from apical regions and lateral
transfer could also be due to a redistribution of the WGA
receptors induced by the binding of the WGA ligand. To
control that the apical membrane still contains the WGA receptors, we sequentially labeled the membrane with WGA conjugated with different fluorochromes. First, we injected WGA–Alexa488 during phase 3, leading to the lateral transfer of that label as described in Fig. 3 A. At the onset of phase 4, WGA–rhodamine was injected in the same region of the embryo. As shown in Fig. 3 D, this second label (red) is predominantly apical and tapers off along the lateral membrane, while the first label (green) is mostly in the basal lateral membrane. These experiments argue that the dynamic changes in WGA are unlikely due to the natural or induced redistribution of the WGA receptors.

In conclusion, throughout the course of cellularization, unlabeled membrane is constantly incorporated into labeled plasma membrane. This incorporation occurs in a defined sequence and a specific pattern, first apical, then apical–lateral. Note that the membrane miscibility appears limited under conditions where WGA is bound to the plasma membrane.

**Displacement of Microspheres on the Membrane Surface**

To further test this idea, we probed the dynamic properties of the plasma membrane in a situation where it is unaffected. We followed the movement of 0.5-μm microspheres bound to an intact plasma membrane. These beads were coated with WGA so they bind the membrane very rapidly upon injection in the vitelline space. The maximum area of contact between the bead and the membrane (0.19 μm²) is much smaller than the total plasma membrane surface area at the onset of phase 3 (at least 100 μm²; see Materials and Methods). Therefore, the beads can be considered as small passive patches representing 1/550th of the total membrane when invagination begins and even less later on. Such beads are thus predicted to follow the global movement of membrane populations they are imbedded in. In particular, if membrane mixing is important and creates a constant homogeneous state in the membrane, the locally delivered new membrane will effectively distribute evenly around the bead.
and, over time, the movement of the bead will be random. However, if membrane mixing is limited, new inserted membrane should consistently displace both the recipient membrane and the beads away from the sites of insertion, thus creating anisotropic growth of the membrane.

We injected WGA-coated fluorescent 0.5-μm beads in the vitelline space of living embryos at different stages. The dilution was such that, on average, less than one bead bound the membrane of a forming cell, to avoid any possible interaction between beads. During phases 1 or 2, beads are seen in the FC a few minutes after injection and remain at the invaginating front through phases 3 and 4. Fig. 4 A shows a bead at time 0 in red and at the end of phase 4 in green (the arrowhead points to the FC). This behavior is observed in 96% of the cases (n = 55) when beads are injected at early stages (Fig. 4 D, top, blue). The remaining 4% represent beads located just above the FC which also accompany the movement of the FC. When injected at the beginning of phase 3, the beads rapidly bind the incipient lateral membrane. Fig. 4 B and B', shows two representative examples where the left panel shows the DIC image at the initial time point: the arrowhead and the small arrows point to the initial positions of the FC and the beads, respectively. The right panel shows the positions of the beads at different time points: the blue bead indicates the initial location (arrows), and subsequent locations are in red and green. The beads are displaced away from the apical sites of membrane insertion and move in register with the advancing FC (Fig. 4 B and B'). 89% of the beads (n = 122) have such a behavior and are eventually located along the basal portion of the lateral membrane (Fig. 4 D, green shaded area). The remaining 11% are found in the medial portion of the lateral membrane (Fig. 4 D, white area). During phase 4, when insertion becomes predominantly apical-lateral, a different situation is observed. Beads injected at the onset of phase 4 remain in the apical region of the membrane, above the insertion site, in 88% of cases (n = 56) (Fig. 4 D, red shaded area) and don’t accompany the rapid movement of the invaginating membrane. Fig. 4 C shows the position of three beads at the end of cellularization; the two arrowheads show the extent of membrane growth during that time window.

Our data show that the microspheres have a reproducible nonrandom mobility and are displaced away from insertion sites with their final distribution depending on the time when they bind the membrane: the most basally located beads were injected earlier than more apically located beads. Given the relatively small size of the beads with respect to the surrounding membrane surface, the movement of the beads most likely reveals the behavior of the supporting membrane as a whole. We therefore infer that membrane growth appears anisotropic, mainly occurring at insertion sites. This situation might reveal that diffusion in the plane of the membrane, although effective, is not enough to cancel the effect of massive membrane insertion in a defined site.

**Insertion of Neurtactin, a Newly Synthesized Transmembrane Protein**

The labeling experiments suggest that membrane growth...

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**Figure 4.** Injection of beads in phase 1/2. (A) Right panel shows a bead at time 0 (red) and the end of phase 4 (green). Left panel shows the DIC image corresponding to the final time point. The arrowhead shows the FC position. (B and B’) Injection in early phase 3. The arrowhead in all images shows the position of the FC at time 0 and the end of phase 4 (end). The DIC image (left) shows the initial time points and the beads (right) are shown in a color-coded fashion: blue at time 0, red later, and green at the end. Note, the beads have accompanied the movement of the FC. The apparent brightness of the beads varies as their position with respect to the confocal optical slices changes during cellularization. (C) Injection in early phase 4. The beads (green) are shown at the end of cellularization together with the DIC image (red). The arrowheads show the relative movement of the FC during the time lapse. (D) Summarizes the observations. Beads are displaced within membrane regions in accordance with the times of injection. Black arrows indicate the sites of membrane insertion.
is mostly accounted for by a reservoir of intracellular membrane originating from the secretory pathway. To assess the contribution of ER and Golgi-based membranes to cellularization, we used antibodies to Neur, a transmembrane protein whose synthesis during cellularization requires de novo RNA synthesis in late syncytial embryos (Fig. 5 A). We followed the localization of Neur along the biosynthetic pathway. Neur is first detected in early phase 2 during nuclear elongation after the basal junction and FCs have formed (Fig. 5 B). At this point, the membrane has not started to grow and most of Neur is localized in punctate structures in the apical and basal cytoplasm (Fig. 5 B, arrowheads). The number and intensity of punctate Neur structures increase in the basal cytoplasm during phase 3 (Fig. 5 C, bracket), suggesting an increased level of synthesis. We compared the cytoplasmic localization of Neur to that of ER and Golgi markers. The ER resident chaperone BiP is concentrated in an apical perinuclear region (Fig. 5 F, arrowhead), and extends at lower levels in an intricate web into the basal cytoplasm (Fig. 5 F, bracket). In contrast to mammalian cells, the Golgi apparatus of insects is dispersed (Ripoche et al., 1994). The cis-Golgi marker βCOP (Oprins et al., 1993; Ripoche et al., 1994) is detected mostly in the basal cytoplasm in punctate structures at the beginning (Fig. 5 G) and the end of cellularization (Fig. 5 H). Therefore, Neur accumulates in a basal region of the cytoplasm where ER and Golgi are found in close association and might exchange vesicles. Neur and βCOP punctate distributions only rarely overlap (Fig. 5 I, arrowheads), although a spatial relationship exists (arrows). This situation may reflect the presence of Neur in regions of the Golgi apparatus devoid of βCOP, or in areas of the ER in close proximity to the cis-Golgi region. We cannot distinguish among these possibilities at present.

During phase 3, Neur also becomes visible along the in-
towards the end of phase 3, Neur levels increase in the lateral membrane, at the expense of a reduced cytoplasmic localization below the nuclei (compare Fig. 5, C and D). It is also detected on the apical surface, but is absent from the FC (marked using an antibody to myosin in Fig. 5 D, inset, red). This latter point is in agreement with our finding that the FC is a membrane region set aside very early before Neur can be detected in the embryo. At the end of cellularization, membrane staining of Neur is even stronger, especially in the apical regions of the lateral membrane (Fig. 5 E, arrowhead). The distribution of Neur is eventually asymmetric, with higher levels coinciding with the zone of late apical–lateral membrane insertion (region 3) observed in WGA labeling experiments. Fig. 5 J shows an embryo labeled with WGA in late phase 3 and viewed at the end of cellularization to illustrate this parallel: the arrowhead points to the zone of unlabeled membrane insertion, which appears to coincide with the zone of increased Neur staining (compare Fig. 5, E and J, arrowheads).

In conclusion, the transmembrane protein Neur is induced during cellularization and localized in the secretory pathway in proximity to cis-Golgi membranes. Neur is continuously inserted to the growing plasma membrane. These observations further support the view that new membrane originating from the secretory pathway is constantly used as raw material for membrane growth.

**Microtubules Mediate the Mobilization of Secretory Membranes to the Growing Plasma Membrane**

Neur must be transported over a large distance of ~20 μm from the Golgi apparatus to the sites of insertion in the plasma membrane. Blocking this transport should immediately stall cellularization if the flux of membrane from the Golgi apparatus is the primary source of new membrane for cellularization. As shown in Fig. 6, a good candidate for the mechanism underlying an apical movement of Golgi membranes is microtubular transport. Microtubules are located apical to the nuclei beneath the plasma membrane (Fig. 6 A, arrowhead) and very long microtubules extend basally up to 30 μm deep inside the embryo (Fig. 6 A and B) well within the area where the bulk of the cis-Golgi membranes are detected (Fig. 6 A) and where Neur accumulates in punctate structures (Fig. 5 C).

Depolymerization of microtubules by injection of colchicine or colcemid at the onset of cellularization blocks membrane invagination (Foe and Alberts, 1983). The effect appears very specific to phases 1, 2, and the beginning of 3, since only a moderate or no reduction in the rate of membrane invagination is detected later in phase 3 and 4 (data not shown). Since Neur is continuously delivered to the growing plasma membrane under normal conditions, we examined the effect of colcemid on Neur distribution. We injected colcemid in embryos at the onset of cellularization, fixed them 30 min later, when cellularization would normally reach completion, and stained them with Neur antibodies. Under these conditions, Neur is virtually absent from the plasma membrane and accumulates at abnormally high levels in punctate structures in the basal cytoplasm reminiscent of those seen under normal conditions (Fig. 6 C). Furthermore, we could not detect any obvious disorganization of the secretory apparatus under these conditions: βCOP is still detected in basal punctate structures (compare Fig. 6, E and A, or Fig. 5 G) and BiP remains in its usual apical location (Fig. 6 D), although we noticed a mild reduction in the basal extent of the BiP signal in colcemid-treated embryos. The exclusion of BiP from the area where Neur accumulates and the similarity between the punctate structures seen with βCOP and Neur (Fig. 6 F) suggest that the Neur punctate structures seen in colcemid-treated embryos are most likely cis-Golgi-associated membranes.

Although we cannot exclude additional roles for microtubules, these data establish a link between plasma membrane growth and export of Neur from Golgi-associated membranes. This requirement further establishes that the membrane needed for cellularization originates from intracellular organelles.

**Discussion**

Using novel techniques to follow the dynamics of the plasma membrane in living embryos, we explored the modalities of membrane growth during cellularization.

We find that cellularization involves the remobilization of an intracellular membrane reservoir and its regulated insertion at well-defined sites in a precise sequence, first apical, then apical–lateral. This unexpected control of membrane delivery during cleavage of the embryo may contribute to the initiation and slow emergence of apical–basal polarity as the epithelium forms.

**Cellularization Involves the Remobilization of an Intracellular Membrane Reservoir**

The origin of the membrane required for cytokinesis and, for cellularization, in particular, has been a long-standing unresolved issue. We conclude that the major reservoir of membrane is intracellular, based on the following observations. First, at any time during cellularization, the previously labeled plasma membrane incorporates membrane that is unlabeled and therefore must originate from inside the cell. Second, Neur, a transmembrane protein that we showed is induced de novo during cellularization, accumulates in Golgi-associated membranes and is continuously transferred to the growing plasma membrane. When microtubules are depolymerized, cellularization immediately stalls and Neur accumulates in abnormally abundant punctate structures of the basal cytoplasm reminiscent of those seen under normal conditions. The exclusion of BiP staining from this region and the presence of βCOP punctate structures in close proximity to colocalizing with Neur argue that Neur is properly exported from the ER to the Golgi apparatus and accumulates in Golgi-associated membranes when microtubules are depolymerized. This is in contrast to mammalian cells, where ER to Golgi export is a microtubule-dependent process and may identify a special feature of the ER and Golgi apparatus organization in early Drosophila embryos. Taken together, our data argue that ER and Golgi membranes constitute the major raw material for plasma membrane growth during cellularization.
cellularization. This is at odds with the idea that membrane grows through the simple redistribution of apical membrane and that the villous projections constitute the major reservoir for invagination (Fullilove and Jacobson, 1971).

Membrane is normally made in the ER and passes through the Golgi apparatus constitutively on its way to the plasma membrane. Given the relatively constant amount of membranes stained with either BiP or βCOP antibodies from cycle 9–14 embryos (not shown), it is probable that the intracellular membrane reservoir is made long before cellularization begins and that cellularization involves the regulated mobilization of this membrane pool to the plasma membrane. It will be interesting to see which steps of the secretory pathway are regulated to accommodate the need for rapid membrane export. Moreover, the shift from a slow rate to a fast rate of invagination suggests that during cellularization regulation of membrane export changes. The existence of a mutant that results in a specific defect in membrane growth during phases 1–3, but where phase 4 (fast phase) proceeds normally (Merrill et al., 1988), supports this view and may provide an entry point into this regulation.

**Insertion of New Membrane as an Active Mechanism during Cytokinesis?**

A surprising result is that the majority of the membrane accounting for growth and invagination is not targeted to the front of the invaginating membrane, which is in contradiction to a report from Loncar and Singer (1995). The appeal of the latter model was that it provided a simple mechanism for directional growth of the membrane. Loncar and Singer (1995) showed electron micrographs where vesicles line up and fuse ahead of the FC, suggesting that the membrane might grow by lateral aggregation of individual vesicles. Our data contradict this scenario. First, Neur is not detected in the FC although its levels build up progressively in the apical and lateral membranes. Second, through our lectin label experiments we show that the FC is a separate membrane region formed in the first 10 min of cellularization (phase 1) and sealed from the adjacent membrane by the presence of a basal junction that accumulates high levels of Arm. Moreover, the membrane is targeted apically in phase 3 or laterally in phase 4 and labeled membrane basal to these insertion sites move basally with the FC. Similarly, microspheres bound to the basal lateral membrane consistently accompany the FC movement. This leads us to conclude that the majority of the new membrane is inserted either apically or apico-laterally, but not at the FC. How might apical or even lateral insertion of membrane contribute to the basal translocation of the FC? Interestingly, our data show that the recipient lateral membrane moves basally in phases 3 and 4 as if pushed away from the insertion point. We propose that insertion of the membrane might play an active role in the invagination process and force the membrane to grow. Directionality could stem from the active role of the contractile actomyosin network that exerts a tension at the tip of the growing membrane (Schejter and Wieschaus, 1993b) or from the pushing force of the microtubules (Foe et al., 1993). However, since in colcemid-treated embryos cellularization proceeds in phase 4 (Foe et al., 1993; our unpublished results), we favor a model in which insertion of the membrane generates the important propulsive force, one that is guided and stabilized by an actomyosin network.
A Link between Membrane Growth and Polarization during Cytokinesis?

Different lines of evidence also suggest that polarization of the plasma membrane might be inherently linked to the process of membrane growth during cleavage of the Drosophila blastoderm. Membrane insertion does not occur at random and uniformly over the entire surface of the plasma membrane. Rather, new membrane is delivered at well-defined sites that change from apical to apical–lateral at different phases of cellularization. This change coincides with a sudden twofold increase in the rate of membrane invagination from phase 3 to 4. Therefore, during cleavage of the Drosophila embryo, membrane trafficking is regulated in a way that allows polarized membrane delivery. Genetic analysis of syntaxin1, a transmembrane protein termed t-SNARE, in Drosophila cellularizing embryos (Burgess et al., 1997) and during cytokinesis of the early Caenorhabditis elegans embryo (Jantsch-Plunger and Glotzer, 1999), have shown that membrane fusion events are required for cytokinesis. However, these studies could not identify where membrane delivery occurred and whether it was polarized. Moreover, it is believed that t-SNAREs do not directly provide spatial cues for membrane delivery since, in budding yeast, the t-SNARE Sec6p/SNA P-25 is localized over the whole cell surface of the forming bud (Brennwald et al., 1994), although membrane delivery only occurs at the tip of the bud. Since then, it has been shown in Saccharomyces cerevisiae (TerBush and Novick, 1995) and in MDCK cells (Grindstaff et al., 1998) that spatial specificity for membrane delivery might be provided by components of a conserved protein complex termed exocyst, and in particular Sec6pSec8 (for a review see Hsu et al., 1999). Sec6 and Sec8 localize to the sites of membrane delivery in contrast to t-SNAREs. Genetic analysis of sec6 and sec8 for which orthologues have been found in Drosophila (Lloyd et al., 2000) might be more revealing than earlier reports on syntaxin. In particular, it should be possible to ask whether the change in the site of membrane insertion from phase 3 to 4 involves the recruitment of D-sec6 and D-sec8 to a new location, or if two independent sets of molecules are involved. In the light of our data, it is tantalizing to propose that the scheme of polarized membrane delivery we identify could participate in the emerging polarization of the plasma membrane.

Two independent sets of experiments argue that membrane miscibility might be insufficient to compete with the effect of massive local membrane delivery, thus lending support to our model. At any time during cellularization, WGA-labeled membrane regions located basal to the sites of membrane delivery move in register with the advancing front of invagination and segregate from the newly inserted membrane as if displaced away from the sites of insertion. We do not think this is a simple effect of WGA binding to the membrane, as a similar conclusion was drawn when we tracked WGA-coated microspheres bound to an otherwise intact plasma membrane. The microbeads serve as landmarks on the plasma membrane and reveal its dynamic properties. Isolated beads move away from the sites of membrane insertion and follow the inward movement of the FC. This situation would not occur if diffusion in the plane of the membrane was enough to cancel the effect of local membrane delivery. Together, these experiments argue that the history of membrane insertion is partly recapitulated in the spatial arrangements of distinct membrane regions, the most basal being also the oldest, generally speaking. This would provide a powerful mechanism to polarize the forming epithelium if the composition of the newly inserted membrane from the ER and Golgi apparatus changes as a function of time through the progressive induction of new transmembrane proteins. Each set of transmembrane proteins might in turn recruit specific groups of membrane-bound proteins and further define each region as a compositionally unique membrane domain. We studied the transmembrane protein Neur and found that its localization in phase 4 reflects the scheme of apico-lateral membrane insertion in agreement with our model. Neur is absent from the FC, a membrane region that is formed before induction of Neur. Neur is also localized at higher levels in the apical–lateral sites of membrane insertion.

Such a model could explain how a prepolarity initiated during cellularization would subsequently participate in recruitment of proteins required for the formation of a mature apical adherens junction. The apical adherens junction is only effective in tightly connecting adjacent cells and serving as a diffusion barrier after cellularization is completed and when gastrulation begins. At this stage, Arm/b-catenin is concentrated in a tight apical belt (Mui ler and Wieschaus, 1996), with E-cadherin, Dlt (Bhat et al., 1999), and Dlg, another PDZ-domain containing protein (Woods et al., 1996; Hough et al., 1997). All these proteins are required for the integrity of the mature adherens junction. Interestingly, the early signs of junction assembly can be tracked back to phase 4 of cellularization when A rm and Dlg become progressively localized to a broad region of the apical lateral membrane (Fig. 1, and our unpublished observations) together with bH- and b-spectrins (Thomas and Williams, 1999). This broad polarization could reflect the insertion of new membrane in this region, as we show (Fig. 3 and 5). Shortly after the completion of phase 4, Dlg and A rm become restricted to the typical apical beltlike localization pattern and Dlt is recruited in the mature junction (our unpublished observations).

To conclude, our data show that cellularization, a specialized form of cytokinesis, is characterized by a precise scheme of membrane growth and insertion that accompanies the emergence of cell polarity. Cellularization may use a novel strategy to polarize the membrane, which we suggest occurs through the sequential insertion of new membrane at different sites and is supported by the fact that membrane mixing between the recipient and newly inserted membrane populations may not be enough to compete with the overwhelming insertion of new membrane. It will be interesting to see whether this pertains to other situations of massive membrane growth such as axon formation. Our findings also open the interesting possibility that membrane insertion might actively participate in furrowing during cytokinesis.

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