AP180 promotes release site clearance and clathrin-dependent vesicle reformation in mouse cochlear inner hair cells

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ABSTRACT
High-throughput neurotransmission at ribbon synapses of cochlear inner hair cells (IHCs) requires tight coupling of neurotransmitter release and balanced recycling of synaptic vesicles (SVs) as well as rapid restoration of release sites. Here, we examined the role of the adaptor protein AP180 (also known as SNAP91) for IHC synaptic transmission by comparing AP180-knockout (KO) and wild-type mice using high-pressure freezing and electron tomography, confocal microscopy, patch-clamp membrane capacitance measurements and systems physiology. AP180 was found predominantly at the synaptic pole of IHCs. AP180-deficient IHCs had severely reduced SV numbers, slowed endocytic membrane retrieval and accumulated endocytic intermediates near ribbon synapses, indicating that AP180 is required for clathrin-dependent endocytosis and SV reformation in IHCs. Moreover, AP180 deletion led to a high prevalence of SVs in a multi-tethered or docked state after stimulation, a reduced rate of SV replenishment and a hearing impairment. We conclude that, in addition to its role in clathrin recruitment, AP180 contributes to release site clearance in IHCs.

This article has an associated First Person interview with the first author of the paper.

KEY WORDS: AP180, SNAP91, Ribbon synapse, Exocytosis, Endocytosis, Hearing, Electron tomography, Membrane capacitance

INTRODUCTION
Ribbon synapses of inner hair cells (IHCs) of the cochlea with spiral ganglion neurons feature one of the highest rates of continuous neurotransmitter release in the mammalian body in order to indefatigably encode sound (Pangršić et al., 2012). Hence, hearing requires, on the one hand, the very fast exocytosis of neurotransmitter-containing SVs and, on the other hand, a highly efficient endocytosis and reformation of fusion-competent synaptic vesicles (SVs) to balance the massive SV exocytosis. The specialized structure of the IHC active zone (AZ) – notably the electron-dense ribbon that is made of RIBEYE (an isoform of CtBP2, herein denoted RIBEYE/CtBP2) and tethers a halo of SVs (Jean et al., 2018; Becker et al., 2018; Kimmich et al., 2005; Schmitz et al., 2000) – and a unique protein composition of release machinery and AZ cytomatrix are thought to accomplish these tasks (reviewed in Moser et al., 2019; Rutherford and Moser, 2016; Safeddine et al., 2012; Wichmann and Moser, 2015). Interestingly, IHC exocytosis seems to operate without the neuronal soluble N-ethylmaleimide-sensitive-factor attachment receptor (SNARE) proteins SNAP25, syntaxin-1 and VAMP2 (Nouvian et al., 2011). Instead, the multi-C2 domain protein otoferlin, loss or dysfunction of which causes human genetic deafness (Yasunaga et al., 1999), serves as a key molecule in IHC exocytosis. After the onset of hearing in mice, otoferlin is thought to be the Ca2+-sensor of exocytosis in IHCs, and also to mediate efficient SV replenishment, possibly by advancing SV tethering and release site clearance (Chakrabarti et al., 2018; Duncker et al., 2013; Johnson and Chapman, 2010; Jung et al., 2015a; Kroll et al., 2019; Michalski et al., 2017; Pangršić et al., 2010; Roux et al., 2006; Strenzke et al., 2016; Vogel et al., 2015, 2016). Clearance of previously exocytosed proteins and lipids from the release sites is considered a critical step in preparing the next round of SV exocytosis (Hauke et al., 2011; Neher and Sakaba, 2008). The action of otoferlin in release site clearance is likely achieved through binding to the adaptor protein 2 (AP-2) (Duncker et al., 2013; Jung et al., 2015a) and/or endophilin A1 (Kroll et al., 2019). Like in neurons (Kononenko et al., 2014), absence of AP-2 from IHCs compromises the clathrin-dependent reformation of SVs from endosomes-like vacuoles (ELVs), but, surprisingly, does not significantly impair endocytic membrane retrieval (Jung et al., 2015a).

Endocytic membrane retrieval in IHCs comprises a slow, linear component that likely reflects clathrin-mediated endocytosis (CME), and a fast, exponential component thought to represent bulk retrieval (Moser and Beutner, 2000; Neef et al., 2014). Morphological evidence for CME and bulk retrieval in hair cells has been reported previously (e.g. Jung et al., 2015a; Lenzi et al., 2002; Neef et al., 2014; Siegel and Brownell, 1986). Molecular entities identified to mediate SV recycling in hair cells include dynamin, endophilin-A1, clathrin and synaptojanin-1 (Boumil et al., 2010; Jung et al., 2015a; Kroll et al., 2019; Neef et al., 2014; Trapani et al., 2009). Their disruption primarily affects the slow component of IHC membrane retrieval, that is, CME. So far, little is known about whether the molecular regulation of bulk retrieval at hair cell synapses is similar to that described for other synapses and neuroendocrine cells (for reviews, see Cousin, 2017; Wu et al., 2014).
A further dissection of the molecular physiology of IHC SV recycling is key to comprehending indefatigable sound encoding. In this context, the neuronal endocytic adaptor protein AP180 is an interesting candidate protein. Through binding of clathrin, AP-2 and the phospholipid phosphatidylinositol 4,5-bisphosphate \([\text{PI}(4,5)P_2]\), AP180 was shown to be involved in the early steps of clathrin-coated pit (CCP) formation in neurons (Ford et al., 2001; Hao et al., 1999; Lindner and Ungewickell, 1992; Morris et al., 1993; Moskhanbaryans et al., 2016). In AP180-knockout (AP180-KO) mice, accumulations of ELVs and lower numbers of clathrin-coated structures have been observed, underlining the importance of AP180 for CME as well as for clathrin-dependent SV reformation from ELVs (Koo et al., 2015). Through its AP180 N-terminal homology (ANTH) domain, AP180 binds to the SNARE domain of VAMP2 and other members of the VAMP family (Koo et al., 2011; Maritzen et al., 2012). This way, AP180, as well as its ubiquitously expressed homolog CALM, organize the recycling of VAMP2 from the plasma membrane after SNARE complex disassembly following SV fusion and, thereby, contribute to the clearance of release sites in neurons (Koo et al., 2011; 2015; Maritzen et al., 2012; Miller et al., 2011). AP180-dependent clearance of release sites seems to be of greater importance in inhibitory neurons that show higher rates of transmitter release than excitatory neurons (Koo et al., 2015). However, the relevance of AP180 for high-throughput synaptic transmission of sensory ribbon synapses is unclear, especially at IHCs, which seem to operate without the AP180 cargo protein VAMP2 (Nouvian et al., 2011).

Here, we studied the role of AP180 at murine IHC ribbon synapses from the molecular to the systems level. By combining confocal microscopy, chemical depolarization of IHCs, high-pressure freezing and electron tomography, perforated patch-clamp, and recordings of otoacoustic emissions and auditory brainstem responses, we discovered that AP180 has a role in release site clearance and clathrin assembly for CME, as well as in SV reformation following bulk retrieval in IHCs, and thereby is important for hearing.

**RESULTS**

**AP180 is present at IHC synapses of the murine cochlea and redistributes upon stimulation**

First, we aimed to identify whether the adaptor protein AP180 is present in the murine organ of Corti, and, more specifically, in IHCs. Therefore, we performed immunohistochemistry in apical cochlear turns (~2–12 kHz) of wild-type (Wt) mice acutely explanted after the onset of hearing, as well as of age-matched AP180-KO mice (used as a control for antibody specificity). We immunostained for AP180 in combination with markers for efferent synapses [synapsin 1 and 2 (synapsin 1/2); note that IHCs do not contain synapsin 1/2], the synaptic ribbon (RIBEYE/CtBP2), and hair cells (otoferlin). Immunofluorescence (Fig. 1A, left panel) revealed the presence of AP180 in IHCs and outer hair cells (OHCS), as well as in efferent synapses underneath the hair cells and possibly in spiral ganglion neurons (see their somata in the inset to Fig. 1A). A lack of AP180 staining in organs of Corti from AP180-KO mice confirmed the high specificity of the AP180 antibody (Fig. S1A). Higher magnifications of maximum intensity z-projections (Fig. 1A, right panel) revealed that AP180 is most prevalent in the basal half of IHCs where the ribbon synapses are located (‘synaptic pole’). Single xy-sections through the synaptic pole of immunolabeled IHCs show an intracellular as well as plasma membrane distribution of AP180. Line profiles through the longitudinal axis of IHCs (Fig. 1B) revealed that the highest fluorescence intensity levels were at the IHC plasma membrane, followed by the IHC lumen and efferent synaptic terminals.

The distribution of AP180 immunofluorescence was altered upon high-K⁺ depolarization (65 mM for 1 min; Fig. S1B). Compared to the resting condition, the average intensity of AP180 fluorescence was significantly reduced in a distance of 2 μm from the plasma membrane (Fig. S1C; \(P<0.0003\), unpaired Student’s \(t\)-test) after high-K⁺ stimulation, whereas the staining near the membrane was preserved (Fig. S1C; \(P=0.5468\), unpaired Student’s \(t\)-test). We further observed that high-intensity AP180 immunofluorescence clusters were already present in the proximity of the plasma membrane at the synaptic pole in the resting condition, but appeared even more intense after stimulation (Fig. S1B). While the size of the clusters did not differ after 1 min of high-K⁺ stimulation compared to in the resting condition (Fig. S1D; \(P=0.105\), Mann–Whitney \(U\)-test), we found a higher mean intensity of AP180 immunofluorescence clusters after stimulation (Fig. S1D; \(P<0.0001\), Mann–Whitney \(U\)-test). These data indicate a stimulation-induced recruitment of AP180 to the plasma membrane, pointing towards a relevance of AP180 for IHC synaptic transmission.

**AP180 is required for normal hearing**

In order to probe for a function of AP180 in IHC synaptic transmission at the systems level, we recorded auditory brainstem responses (ABRs) and distortion product otoacoustic emissions (DPOAEs). Wt and AP180-KO mice showed comparable ABR wave amplitudes (Fig. 2A,D) and ABR latencies (Fig. 2E), while the ABR thresholds were modestly but significantly increased in the AP180-KO mice (Fig. 2B; \(P<0.01\), two-way ANOVA followed by post-hoc Tukey’s test). DPOAEs, reflecting cochlear amplification, were intact in AP180-KO mice, which reveals normal mechanoelectrical transduction and electromotility of OHCs (Fig. 2C). Therefore, we conclude that the elevation of ABR thresholds arises from a dysfunction downstream of mechanoelectrical transduction and cochlear amplification, that is, in IHCs, their synapses or the auditory pathway.

**AP180 absence alters the levels of Vglut3, but not of otoferlin**

In IHCs of mice with a knockout of the AP-2 subunit AP-2\(\mu\) (also known as AP2M1), otoferlin levels are known to be strongly reduced, with the reduction being less pronounced at the plasma membrane, suggesting impaired sorting of otoferlin (Jung et al., 2015a). Therefore, we tested by semi-quantitative analysis of otoferlin immunofluorescence if deletion of AP180 likewise changes otoferlin levels and/or distribution (Fig. 3A,A). Yet, neither otoferlin distribution, nor total fluorescence intensity, were altered in IHCs of AP180-KO mice compared to Wt (\(P>0.17\), unpaired Student’s \(t\)-test). However, we noted that the immunofluorescence intensity of the vesicular glutamate transporter of IHCs, Vglut3 (also known as SLC17A8), was reduced by 17.1±3.2\% (mean±s.e.m.) throughout the IHC in AP180-KO mice compared to Wt (Fig. 3B; \(P<0.0001\), unpaired Student’s \(t\)-test). However, quantitative PCRs of AP180-KO and control (Wt and heterozygote) mice [postnatal day (P)12–P14] revealed that mRNA levels of otoferlin and Vglut3 remained unchanged (Fig. S2). Notably, co-staining for RIBEYE/CtBP2 and Homer1 (a marker for the postsynaptic density; Fig. 3C) indicated an unaltered ribbon anchorage to the AZ as well as a normal number of ribbon synapses (Fig. 3C; ribbon synapses per IHC for Wt: 14.5±0.4, for AP180-KO: 14.5±0.5 synapses per IHC; \(P=0.96\), unpaired Student’s \(t\)-test; mean±s.e.m.) in AP180-KO mice. In summary, deletion of AP180 did not alter otoferlin levels, synapse number or ribbon anchorage, but it reduced Vglut3 protein levels, potentially via diminishing SV numbers.
The numbers of SVs and clathrin-coated structures are reduced in IHCs of AP180-KO mice

Next, we performed high-pressure freezing followed by freeze substitution and electron tomography of ribbon synapses from P15–P16 mice (shortly after the onset of hearing) to assess the synaptic ultrastructure. Exemplary virtual sections (Fig. 4A), obtained from semi-thin sections via tomogram generation, as well as 3D models of reconstructed ribbon synapses from Wt and AP180-KO IHCs (Fig. 4B) revealed reduced numbers of SVs not only in direct vicinity to the ribbon, but also in the cytoplasm. We investigated the following three morphological pools of SVs, as defined previously (Kroll et al., 2019) and as indicated in the schematic drawing (Fig. 4D): (1) ribbon-associated SVs (RA-SVs) in a maximum distance of 80 nm to the ribbon, (2) membrane-proximal SVs (MP-SVs) that are within a 50 nm distance from the plasma membrane and 100 nm laterally from the presynaptic density, and (3) cytosolic SVs (operationalized as all uncoated vesicles with a maximum outer diameter of 70 nm and a maximum distance of 500 nm from the ribbon, excluding the aforementioned SV pools). In all three pools, numbers of SVs were reduced in IHCs of AP180-KO mice compared to Wt. The strongest reduction, of ~60%, was observed for the pool of cytosolic SVs (Fig. 4I; $P<0.0001$, unpaired Student’s $t$-test). Numbers of RA-SVs (Fig. 4E; $P=0.0062$, unpaired Student’s $t$-test) and of MP-SVs (Fig. 4F; $P=0.0328$, unpaired Student’s $t$-test) were reduced by ~24% and 22%, respectively. The reduction of SVs, found in electron tomography, exceeded the expectation based on Vglut3 immunofluorescence (reduction by ~17%). The outer diameter of the cytosolic SVs, on the other hand, was increased on average (Fig. 4J; $P<0.0001$, unpaired Student’s $t$-test) and more variable in IHCs of AP180-KO mice compared to Wt ($P<0.0001$, Kolmogorov–Smirnov test), which may account for the modest reduction of Vglut3 fluorescence intensity. The observed increase of the SV diameter is consistent with results of previous studies on mice and invertebrates missing AP180 or its homolog UNC-11 (Koo et al., 2015; Nonet et al., 1999; Vanlandingham et al., 2014; Zhang et al., 1998). Interestingly, the average distance of MP-SVs to the AZ plasma membrane was slightly increased in IHCs from AP180-KO mice compared to Wt ($P=0.0288$, Mann–Whitney U-test).

We further quantified the numbers of clathrin-coated structures in proximity to the ribbon, including coated vesicles and coated pits budding from the plasma membrane or from ELVs, examples of which can be seen in Fig. 4C. The numbers of coated structures in total (Fig. 4H; $P=0.0002$, unpaired Student’s $t$-test) and of coated vesicles (Fig. 4I; $P=0.0062$, unpaired Student’s $t$-test) were reduced by ~60%, and ~50%, respectively, in IHCs of AP180-KO mice compared to Wt. In contrast, the numbers of coated pits budding from the plasma membrane or from ELVs were not significantly different between Wt and AP180-KO IHCs (Fig. 4J; $P=0.0541$, unpaired Student’s $t$-test).
t-test), and of coated vesicles in particular (P<0.0001, Mann–Whitney U-test, please also see Table S1 for further information on statistical analyses), were strongly reduced in AP180-KO mice compared to Wt. This suggests an involvement of AP180 in the early steps of clathrin-mediated membrane retrieval. While numbers of ELVs were not significantly increased in tomograms compared to Wt. This suggests an involvement of AP180 in the early steps of clathrin-mediated membrane retrieval. While numbers of ELVs were not significantly increased in tomograms.

![Fig. 2. Auditory brainstem responses show elevated thresholds in AP180-KO mice. (A) ABR waveforms were recorded in response to 80 dB clicks (mean±s.e.m., AP180-KO N (number of mice)=9, Wt N=9). Roman numerals (I–V) indicate the ABR waves. (B) ABR thresholds were significantly elevated for AP180-KO mice compared to Wt mice (mean±s.e.m., AP180-KO N=9, Wt N=9). **P<0.01 (two-way ANOVA followed by post-hoc Tukey’s test). Click thresholds (for 20 and 100 Hz) remained unchanged. (C) DPOAE amplitudes (mean±s.e.m.) recorded in response to the pair of simultaneous sine waves (f1=13.3 kHz and f2=16 kHz) at increasing intensity (f2 intensity was 10 dB bigger than f1 intensity) were unaltered. (D) The cumulative amplitude from wave 1 to 5 was comparable in AP180-KO and Wt mice. (E) Latencies of ABR waves I–V did not show any difference in AP180-KO mice compared to Wt mice. Results from individual mice are shown as fainter lines in C and D.]

![Fig. 3. Reduced Vglut3 levels, but unaltered otoferlin levels in AP180-deficient IHCs. (A) Maximum intensity z-projections of otoferlin-stained apical cochlear turns from AP180-KO and Wt littermate controls. (A) Unaltered otoferlin levels in AP180-KO (n=114 IHCs from N=4 mice) compared to Wt (n=107 IHCs from N=4 mice). (B) Maximum intensity z-projections of confocal sections of Wt and AP180-KO IHCs stained for Vglut3. (B) Intracellular Vglut3 levels were reduced by ~17% in AP180-KO IHCs (n=114 IHCs from N=4 mice) compared to Wt (n=107 IHCs from N=4 mice). (C) Exemplary confocal sections of RIBEYE/CtBP2 (magenta) labeling the synaptic ribbon and Homer1 (green) staining labeling the postsynaptic density. Dashed circles highlight the nuclei. (C) Equal numbers of ribbon synapses in Wt (six images from three animals, with ~10 cells per image) and in AP180-KO (six images from three animals, with ~10 cells per image). ***P<0.001. Scale bars: 5 μm.]
of AP180-KO mice (Fig. 4K; \(P = 0.7645\), unpaired Student’s \(t\)-test), we found a strong increase in their average volume (Fig. 4L; \(P = 0.0043\), Mann–Whitney \(U\)-test). Therefore, it is likely that the reduced numbers and increased size of SVs are a consequence of impaired clathrin- and AP180-dependent SV reformation from ELVs.
IHCs from AP180-KO mice show normal Ca²⁺ influx but impaired SV replenishment

In order to test for the physiological correlates of the morphological changes observed in the AP180-KO IHCs, we performed perforated patch-clamp experiments soon after the onset of hearing (5 mM [Ca²⁺]o, P14–P18) in acutely explanted apical cochlear coils (∼6–10 kHz). We recorded whole-cell Ca,1.3-mediated Ca²⁺ influx in response to step depolarizations of 10 ms from −84 mV to +63 mV in 5 mV increments (Fig. 5A, inset). The amplitude of the Ca²⁺ influx of AP180-KO IHCs was statistically indistinguishable from that of Wt IHCs (Fig. 5A). Furthermore, we performed measurements of membrane capacitance increments (ΔCm), which reports exocytosis evoked by the Ca²⁺ influx. SV pool dynamics were probed by a train of stimuli that consisted of ten step depolarizations to −14 mV of 20 ms duration each, with 150 ms inter-stimuli intervals (Fig. 5B). No significant differences of ΔCm in response to the first depolarization, which is thought to represent fusion of the readily releasable pool (RRP) of SVs, or in response to the full train of stimuli, were observed (Fig. 5B1, B2). We further examined the replenishment rate of the SVs by using the ‘SMN analysis’ (Schneggenburger et al., 1999). We plotted the cumulative ΔCm versus the stimulus number and fitted a line for the final five stimuli (Fig. 5C). The obtained apparent replenishment rate (ΔfF/stimulus) was significantly lower in AP180-KO IHCs (Fig. 5Ci, P=0.037, unpaired Student’s t-test). The increased SV size observed in AP180-KO mice leads to an overestimation of the true rate of SV replenishment. Assuming a spherical shape and a specific capacitance of 9.1 fF/μm² (Albillos et al., 1997), we estimated the expected capacitance added by single SV fusion (SV diameter was obtained from the tomograms; Fig. 4J). The mean estimated outer SV diameter values were 44.0±0.1 nm and 48.4±0.3 nm (mean±s.e.m.), for Wt and AP180-KO IHCs, respectively. We subtracted the average membrane thickness (6.2 nm, Neef et al., 2007) to obtain the mid-membrane diameter, which is relevant for calculating the capacitance increase upon single SV fusion. The estimated capacitance for Wt SVs (41.3±0.3 aF) was close to the measured value of 40 aF (Grabner and Moser, 2018), while the SV capacitance predicted for AP180-KO IHCs amounted to 51.8 ±0.7 aF. Based on these estimates, we converted the apparent SV replenishment rate (ΔfF/stimulus, see above) into number of vesicles per stimulus. This revealed a strong impairment in the SV replenishment rate in AP180-KO IHCs (Fig. 5Cii, P=0.001, unpaired Student’s t-test). Potential reasons for the observed reduction of the replenishment rate in AP180-KO IHCs include (1) deficits in SV docking and/or priming, (2) disturbed release site clearance and (3) impaired SV resupply, including SV reformation and recruitment of SVs to the AZ, which we aimed to decipher in the following experiments.

Deletion of AP180 impairs CME and clathrin-dependent SV reformation in IHCs

Given the role of AP180 in CME and clathrin-dependent SV reformation at conventional synapses (Koo et al., 2015; Zhang et al., 1998), we investigated endocytic membrane retrieval in IHCs of AP180-KO mice. By performing perforated-patch recordings, we determined the endocytic Cm decline following exocytosis in response to step depolarizations to −14 mV for 20 ms and for 200 ms (Neef et al., 2014). As above, both AP180-KO and Wt IHCs showed comparable exocytic ΔCm upon 20 ms and 200 ms step depolarizations (Fig. 6A1, B1). The slow and linear component of endocytosis, likely to reflect CME (Neef et al., 2014), dominated the endocytic Cm decline following 20 ms depolarizations in IHCs of both genotypes (Fig. 6C1). The slope of the linear component was significantly smaller in AP180-KO IHCs compared to Wt IHCs, indicating a deficit in CME (Fig. 6F, P=0.043, Mann–Whitney U-test). In contrast, the slope of the linear component was comparable between AP180-KO and Wt IHCs upon 200 ms step depolarization (Fig. 6G). Furthermore, AP180 deletion did not impair the fast, exponential component of the endocytosis elicited by exocytosis in response to 200 ms depolarization (Fig. 6D1, H1), which likely reflects bulk endocytosis (Neef et al., 2014).

In a next step, we stimulated freshly dissected P15–P16 organs of Corti for 5 min using high-K⁺ followed by high-pressure freezing, freeze substitution and electron tomography (Fig. 7). Similar to what is found in resting conditions, we observed a lower total number of coated structures in IHCs of AP180-KO mice (Fig. 7I, P=0.002, Mann–Whitney U-test). We detected several ELVs with multiple budding CCPs in proximity to ribbon synapses in Wt, indicative of active SV reformation following bulk endocytosis (examples are shown in the virtual section and in the 3D model in Fig. 7C). However, in AP180-KO mice, we did not observe ELVs with more than one or two budding CCPs. Moreover, the number of ELVs with one or more budding CCPs was reduced in AP180-KO IHCs compared to Wt (Fig. 7I; P=0.006, Mann–Whitney U-test, also see Table S1). As seen at rest, the total number of ELVs was unaltered in AP180-KO IHCs compared to Wt (P=0.113, Student’s t-test; Table S1) whereas the average ELV volume was again increased (Fig. 7H; P=0.0355, Mann–Whitney U-test).

The number of cytosolic SVs was decreased by more than 40% in AP180-KO IHCs (Fig. 7F; P=0.0026, Mann–Whitney U-test), and, hence, did not go beyond the reduction found at resting conditions. Interestingly, we neither observed a difference in the number of RA-SVs (Fig. 7D; P=0.445, Mann–Whitney U-test) nor in the number of MP-SVs (Fig. 7E; P=0.721, Mann–Whitney U-test) between Wt and AP180-KO mice after stimulation. These data indicate that AP180- and clathrin-dependent reformation of SVs is required for maintaining high numbers of cytosolic SVs in the synaptic pole of the IHC. Still, even in absence of AP180, endocytic SV recycling was sufficient to provide enough SVs for refilling the pools of RA-SVs and MP-SVs. Therefore, the role of AP180 in CME and SV reformation seems unlikely to fully account for the observed impairment in SV replenishment.

After stimulation, SVs remain in a multi-tethered state at the AZ of AP180-KO IHCs

In order to investigate potential mechanisms underlying impaired SV replenishment, we further examined the MP-SV pool by electron tomography (Fig. 7A1, B1). As mentioned above (Fig. 4G), the distance of MP-SVs to the AZ membrane was increased in AP180-KO IHCs under resting conditions. After stimulation, we did not observe significant differences in the average distance to the AZ membrane for MP-SVs between Wt and AP180-KO IHCs (Table S1; P=0.451, unpaired Student’s t-test). Since SV tethering to the AZ membrane has been suggested to influence the distance between MP-SVs and AZ membrane (e.g. Chakrabarti et al., 2018; Fernández-Busnadiego et al., 2013), we quantified the numbers of tethers to the AZ membrane under resting conditions and after stimulation. For that purpose, we defined the following SV groups: (1) SVs without such tethers, (2) SVs with a single tether, (3) SVs with multiple tethers, and (4) docked SVs, where the distance between SV outer membrane and AZ membrane was 0–2 nm in electron tomography virtual sections, as previously described for IHCs and neurons (Chakrabarti et al., 2018; Imig et al., 2014).
In resting conditions, we did not observe any significant differences in the tethering of MP-SVs in AP180-KO IHCs compared to what was found in the Wt situation (Fig. 7G, one-way ANOVA followed by Tukey’s post-hoc test; for individual P-values see Table S1). In line with a previous study of IHCs, high-K+ stimulation resulted in relatively more tethered SVs (Chakrabarti et al., 2018). This was the case in both Wt and AP180-KO IHCs (Fig. 7G; for the fraction of untethered SVs, resting Wt versus stimulated Wt P=0.001, resting Wt versus stimulated AP180-KO P=0.001, one-way ANOVA followed by Tukey’s post-hoc test). However, we noticed a shift towards an increase in multi-tethered SVs in AP180-KO IHCs after stimulation, whereas most SVs in stimulated Wt IHCs remained connected via a single tether (P=0.008 for the fraction of multi-tethered MP-SVs in stimulated Wt versus stimulated AP180-KO IHCs). Furthermore, we observed significantly more docked SVs in stimulated AP180-KO IHCs compared to resting conditions (Wt resting condition versus AP180-KO stimulated condition: P=0.004, one-way ANOVA followed by Tukey’s post-hoc test; AP180-KO resting versus AP180-KO stimulated: P=0.002, one-way ANOVA followed by Tukey’s post-hoc test), as well as compared to stimulated Wt (P=0.037, one-way ANOVA followed by Tukey’s post-hoc test). Together, these observations indicate that MP-SVs in IHCs of AP180-KO mice remain in the multi-tethered or docked state prior to fusion, which has previously been designated as a morphological correlate of impaired release site clearance in IHCs (Chakrabarti et al., 2018).

DISCUSSION

In the present study, we addressed a role of the endocytic adaptor protein AP180 in synaptic sound encoding in the cochlea. There, IHC ribbon synapses must sustain high rates of SV release for reliable transmission of auditory information. When investigating IHC ribbon synapses of AP180-KO mice by performing a combination of functional patch-clamp recordings, electron tomography and systems physiology, we found evidence for a dual function of AP180. Firstly, AP180 is required for clathrin-dependent SV reformation from ELVs and aids in the early steps of classical CME. Secondly, AP180 plays a role in efficient replenishment of SVs to the release site, likely via promoting the clearance of the release site from proteins and lipids added by preceding SV fusion events.
AP180 is required for clathrin-dependent endocytosis and SV reformation following bulk retrieval in IHCs

The reduced rate of endocytic C_m decline and the reduction of clathrin-coated SVs in resting and stimulated AP180-KO IHCs support a role of AP180 in CME. Beyond that, we observed less budding of CCPs from enlarged ELVs in AP180-KO IHCs after high-K+ stimulation, suggesting that SV reformation from ELVs following bulk retrieval operates in an AP180-dependent manner.

Clathrin-dependent SV reformation from ELVs was also impaired in AP-2 KO IHCs (Jung et al., 2015a). Therefore, like in hippocampal neurons (Kononenko et al., 2014; Koo et al., 2015), both adaptor proteins seem to be required for the recruitment of clathrin triskelia not only to the plasma membrane, but also to ELVs. Notably, SV numbers were more robustly reduced in AP180-deficient IHCs (∼60%) than in AP180-deficient inhibitory CNS neurons (∼33%; note that no SV reduction was found in excitatory neurons, Koo

Fig. 6. A reduced rate of the slow component of endocytic membrane retrieval, but intact fast endocytosis in AP180-KO IHCs. (A) Exemplary traces of Ca^{2+} current (top) and capacitance (bottom) upon 20 ms step depolarization to −14 mV from the holding potential of −84 mV. AP180-KO IHCs had comparable ΔC_m (Ai) and efficiency of Ca^{2+} current to drive exocytosis, as probed by the ratio of ΔC_m and Q_{Ca} (Ai), in response to 20 ms step depolarization, primarily eliciting RPP release (AP180-KO n=15 IHCs, N=12 mice; Wt n=15 IHCs, N=11 mice). (B) Exemplary traces of Ca^{2+} current (top) and capacitance (bottom) upon 200 ms step depolarization. ΔC_m in AP180-KO IHCs was comparable with Wt IHCs in response to 200 ms depolarization eliciting sustained release (Bi). The efficiency of Ca^{2+} current to drive exocytosis was comparable (Bii). Average traces of C_m in response to 20 ms (C) and 200 ms (D) depolarization (means±s.e.m., AP180-KO n=16 IHCs, N=12 mice; Wt n=17 IHCs, N=14 mice). (E) The time of return to baseline after 20 ms depolarization was comparable between AP180-KO and Wt IHCs. (F) The slope of the slow linear component of endocytic membrane retrieval, most likely reflecting CME, was calculated by fitting a line to the first 20 s of the C_m recording after the depolarization. AP180-KO IHCs showed a shallower slope (without the outliers, *P=0.002, Student’s t-test, with the outliers, *P=0.043, Mann–Whitney U-test). (G) The slope of the linear component following 200 ms depolarization was not statistically distinguishable. We did not observe significant differences in the amplitude (H) and the time constant (I) of the fast, exponential component of endocytosis, likely to represent bulk endocytosis. The outliers were detected via inter-quartile range (IQR) method and were depicted as filled diamonds. *P<0.05; ns, not significant.
et al., 2015). This underlines the importance of clathrin- and AP180-dependent SV reformation from endocytic intermediates for the restoration of SV pools particularly in cells with a high SV turnover rate like the IHCs.

Nonetheless, CME still occurred, and bulk retrieval of membrane seemed unaffected in AP180-deficient IHCs. Moreover, prolonged stimulation by high-K⁺ left the RA- and MP-SVs pools unchanged and did not further reduce the number of cytosolic SVs in AP180-KO IHCs. Therefore, AP180-independent forms of membrane retrieval must exist in IHCs, and other adaptor proteins, like the AP180 homolog CALM, AP-2 or AP-3, may contribute to the recruitment of clathrin. The precise interplay of adapter proteins and membrane retrieval mechanisms in IHCs requires further investigation. Also, future studies might address the question of whether accumulation of exocytosed proteins and lipids at the plasma membrane, or increased membrane tension, shift membrane retrieval towards clathrin-independent forms (Maritzen and Haucke, 2018).

Fig. 7. SV tethering to the AZ and SV reformation are altered in stimulated IHCs from AP180-KO mice. (A) Reconstructed models of the AZ from P15 Wt and AP180-KO IHCs after 5 min K⁺ stimulation showing the presynaptic density and MP-SVs. Colors of different classes of MP-SVs are as highlighted in B. Scale bars: 200 nm. (B) Exemplary virtual sections of MP-SVs in untethered, tethered or docked state. Scale bars: 50 nm. (C) Virtual section and 3D model of an ELV containing multiple CCPs, as exclusively observed in Wt. Scale bars: 100 nm. (D) Numbers (N) of RA-SVs were unaltered in P15 AP180-KO mice compared to Wt (n for D-I=10 tomograms from two different animals per group). (E) The number of MP-SVs was likewise unaltered in absence of AP180 compared to Wt, whereas numbers of small uncoated vesicles were strongly reduced (F). (G) Examining numbers of tethers, a significantly higher proportion of multi-tethered and docked MP-SVs have been observed combined with a relatively lower number of single-tethered SVs in absence of AP180 after stimulation compared to stimulated Wt as well as compared to resting condition in Wt and AP180-KO. (H) The volume of ELVs was increased in the AP180-KO mice. (I) Numbers of coated structures were decreased with numbers of coated vesicles and coated pits at ELVs being most strongly affected. *P<0.05; **P<0.01; ns, not significant.

A role of AP180 in release site clearance at IHC active zones

Notwithstanding the SV reformation deficit, the number of MP-SVs at the AZ was not significantly reduced in AP180-KO IHCs during strong stimulation. Still, the measured rate of SV replenishment during trains of brief depolarizations was reduced in AP180-KO IHCs, while Ca²⁺-triggered fusion of the RRP of SVs seemed intact. These findings suggest that slowed SV replenishment of the release sites was primarily due to impaired AZ clearance rather than to insufficient SV resupply to the AZ. Effective release site clearance has previously been discussed as a determinant of sustained exocytosis in hair cells (Pangršić et al., 2010; Cho et al., 2011; Jung et al., 2015; Chakrabarti et al., 2018; Kroll et al., 2019). Further support for this hypothesis comes from the observation of an increased number of multi-tethered and docked SVs in the AP180-KO IHCs. Similarly, in IHCs of the otoferlin mouse mutant pachanga, which has been shown to be profoundly deaf with a reduced rate of SV replenishment to the RRP (Pangršić et al., 2010), the number of multi-tethered SVs was significantly higher than in Wt
after stimulation (Chakrabarti et al., 2018). Multi-tethered SVs in neurons were also shown to be closer to the AZ membrane compared to single-tethered or untethered SVs, and probably display a prerequisite for SV docking and priming (Fernández-Busnadiego et al., 2013). As SV fusion was intact in AP180-KO IHCs, we take the higher prevalence of multi-tethered SVs as a potential morphological correlate of unproductive release sites due to impaired clearance.

In a previous study, absence of RIM-binding protein 2 (RIM-BP2) likewise resulted in an increased distance of MP-SVs to the AZ-membrane in IHCs at resting conditions, which has been attributed to disturbances in the composition of the cytomatrix of the AZ (Krinner et al., 2017). Changes in the cytomatrix or in the AZ plasma membrane itself may also serve as an explanation for the increased distance of MP-SVs in absence of AP180. More precisely, the absence of AP180 could lead to an accumulation of proteins or lipids at the membrane of the release sites, which possibly causes an energetically unfavorable environment, ultimately slowing down exocytic processes after prolonged stimulation.

AP180 has previously been identified as a key sorting factor for VAMP2. Consequently, AP180 deficiency causes an accumulation of VAMP2 at the plasma membrane of neurons (Koo et al., 2015). Even though IHCs seem to operate without VAMP2 and other neuronal SNARE proteins (Nouvian et al., 2011), AP180 could be required for the sorting of other members of the VAMP family that were previously identified as additional interaction partners of AP180 or CALM (Maritzen et al., 2012; Miller et al., 2011; Sahlender et al., 2013). Alternatively, an as-yet-unknown exocytic IHC protein may interact with AP180 and accumulate at the plasma membrane in the absence of AP180. Importantly, unaltered otoferlin levels suggest that otoferlin sorting does not depend on AP180 and consequently allows us to attribute the IHC phenotype to AP180 deficiency itself rather than to a concomitant loss of otoferlin.

**MATERIALS AND METHODS**

**Animals**

Constitutive knockout mice for AP180 as well as their wild-type littermates, as previously described in Koo et al. (2015), were used in this study. For all experiments, mice of either sex were examined shortly after the onset of hearing (P14-P18). For the analysis of AP180 localization and distribution, C57BL/6J mice were used. All experiments complied with national animal care guidelines and were approved by the University Medical Center Göttingen board for animal welfare and the animal welfare office of the state of Lower Saxony.

**Immunohistochemistry and confocal microscopy**

Freshly dissected apical turns of organs of Corti from 2-week-old mice were (1) chemically stimulated for 1 min using high-K" +" stimulation solution (65.36 mM KCl, 79.7 mM NaCl, 2 mM CaCl_2, 1 mM MgCl_2, 0.5 mM MgSO_4, 10 mM HEPES, 3.4 mM L-glutamine, and 6.9 mM D-glucose, pH 7.4) or incubated in a high Ca" +" solution for 2 h (5.63 mM KCl, 139.7 mM NaCl, 2 mM CaCl_2, 1 mM MgCl_2, 0.5 mM MgSO_4, 10 mM HEPES, 3.4 mM L-glutamine, and 6.9 mM D-glucose, pH 7.4; see also Revelo et al., 2014) at room temperature followed by fixation using 4% formaldehyde (FA) in phosphate-buffered saline (PBS) on ice, or (2) were directly fixed with 4% FA in PBS for 10 min (for RIBEYE/CtBP2 and Homer1) or for 60 min (for otoferlin, Vglut3 and AP180) on ice. After three 10 min washing steps in PBS, blocking solution [goat serum dilution buffer (GSDB), 1% normal goat serum, 450 mM NaCl, 0.3% Triton X-100, 20 mM phosphate buffer, pH 7.4] was applied for 1 h in a wet chamber at room temperature. Primary antibodies were diluted in GSDB and applied overnight at 4°C in a wet chamber. After three 10 min washes (wash buffer, 450 mM NaCl, 20 mM phosphate buffer, 0.3% Triton X-100), secondary antibodies diluted in GSDB were applied in a light-protected wet chamber for 1 h at room temperature. Then, the specimens were washed three times in wash buffer and finally in 5 mM phosphate buffer and mounted onto glass microscope slides with mounting medium (Mowiol 4-88, Sigma-Aldrich).

The following primary antibodies were used: rabbit anti-AP180 (1:300, Synaptic Systems, cat. No. 155 003), mouse anti-CtBP2 (also recognizing the ribbon protein ribeye, 1:200, BD Biosciences, cat. no. 612044), mouse anti-otoferlin (1:300, Abcam, cat. no. ab53233), rabbit anti-Vglut3 (1:300, Synaptic Systems, cat. no. 135 203), rabbit anti-Homer1 (1:200, Synaptic Systems, cat. No. 160 002) and guinea pig anti-synapsin1/2 (Synaptic Systems, cat. no. 106 002). The secondary antibodies used for confocal microscopy were goat anti-rabbit-IgG conjugated to Alexa Fluor 488 (1:200, Invitrogen, cat. No. A11008), goat anti-mouse-IgG conjugated to Alexa Fluor 568 (1:200, Invitrogen, cat. No. A11004), goat anti-mouse-IgG conjugated to Alexa Fluor 633 (1:200, Invitrogen, cat. no. A21136), and goat anti-guinea pig-IgG conjugated to Alexa Fluor 568 (1:200, Invitrogen, cat. No. A11075).

Confocal images were acquired using a laser scanning confocal microscope (Zeiss LSM800, Carl Zeiss AG, Oberkochen, Germany) with 488-nm (Ar) and 561 nm (He-Ne) lasers for excitation and 1.4 NA 63× oil immersion objectives. z-axis stacks of 2D images were generated with a step size of 0.6 μm from comparable tonotopic regions. Images were processed using ImageJ (http://imagej.net/) and assembled for display in Adobe Illustrator software. For semiquantitative analysis of immunofluorescence, samples of AP180-KO and control/unstimulated and stimulated probes were processed identically and in parallel throughout immunohistochemistry, confocal imaging and analysis.

**Quantitative RT-PCR**

Total RNA was isolated from the cochlea of 12–14-day-old mice using the PureLink™ RNA Mini Kit in combination with Trizol (Thermo Fisher Scientific). The SuperScript® IV First-Strand Synthesis System (Thermo Fisher Scientific) was used to obtain cDNA according to manufacturer’s instructions using oligo(dT) primers and random hexamers. Quantitative RT-PCR (qPCR) was performed using a PowerUP SYBR™ Green master mix and gene-specific primers for Vglut3 and otoferlin (see Table S2 for primer sequences). The StepOne™ Real-Time PCR System (Thermo Fisher Scientific) was used to perform qPCR. Each PCR was repeated three times in one experimental run.

**High-pressure freezing and freeze substitution**

High-pressure freezing (HPF) and freeze substitution (FS) were essentially performed as described previously (Chakrabarti et al., 2018; Jung et al., 2015b; Wong et al., 2014). Briefly, for HPF, the apical cochlear turns from P15–P16 Wt and AP180-KO mice were dissected in Ca" +"-free Hanks’ balanced salt solution (HBSS) and carefully placed on aluminum specimen carriers in the 0.2 mm cavity (type A, 0.1 and 0.2 mm cavity, Leica Microsystems, Wetzlar, Germany) filled with HBSS solution. A second specimen carrier (0.3 mm cavity, type B, Leica Microsystems) was dipped in hexadecane and placed onto the first specimen carrier with the cavity upwards. An HPM100 (Leica Microsystems) high-pressure freezer was used for sample freezing. Immediately afterwards, samples were transferred into liquid nitrogen and stored until freeze substitution was performed. For stimulation, organs of Corti were dissected in Ca" +"-free HBSS solution and transferred into stimulation solution (identical with the solution used for immunohistochemistry experiments). Within the stimulation solution, samples were placed on specimen carriers and frozen exactly 5 min after the transfer.

Freeze substitution was performed with an AFS2 (Leica Microsystems) freeze substitution device. Organs of Corti were transferred into the AFS2 precooled to −90°C. After 4 days incubation in 0.1% tannic acid in aceticone, samples were washed three times for 1 h each time in aceticone before 2% (v/v) osmium tetroxide in aceticone was applied at −90°C. The temperature was increased from −90°C to −20°C (5°C/h), maintained at −20°C for 17 h and was further increased from −20°C to +4°C (10°C/h). Osmium tetroxide was removed at 4°C and samples were washed three times for 1 h each time with aceticone. Subsequently, samples were slowly warmed to room temperature. Finally, organs of Corti were infiltrated with Epon resin [acetohe/Epon 1:1 (v/v) for 2 h, 100% Epon overnight], placed into embedding molds and polymerized for 48 h at 70°C.

**Electron tomography**

Electron tomography of ribbon synapses was performed as described previously (Jung et al., 2015b; Strenzke et al., 2016). 250 nm sections of the
embedded samples were obtained, approaching from the anterior edge, using an ultramicrotome (UC6, Leica Microsystems, Wetzlar, Germany) with a 35° diamond knife (Diatome, Didau, Switzerland) and applied to formvar-coated copper 75-mesh grids. Sections were post-stained with uranyl acetate replacement solution (Science services, Munich, Germany) for 40 min and lead citrate for 1 min following standard protocols. To both sides of the grids, 10 nm gold beads (British Bio Cell, Crumlin, UK) were applied as fiducial markers. Using Serial-EM software, single tilt series from −60° to +60° (increments of 1°) were acquired with a JEM2100 (JEOL, Freising, Germany) electron microscope at 200 kV and 10,000× magnification. For tomogram generation, the IMOD package etomo was used; models were generated with 3dmod (bio3d.colorado.edu/imod/).

### Auditory brainstem recordings and distortion product otoacoustic emissions

ABRs and DPOAE were performed on 6-week-old mice as previously described (Jing et al., 2013). Briefly, mice were anesthetized with a combination of ketamine (125 mg/kg body weight) and xylazine (2.5 mg/kg body weight) through intraperitoneal injection. The temperature was maintained constant at 37°C using a heat blanket (Schno-Sachs Elektronik–Harvard Apparatus). The TDT II system run by BioSig software was used for stimulus generation, presentation and data acquisition (Tucker Davis Technologies; MathWorks). Tone bursts (as a sequence at 4, 6, 8, 12, 16, 24 and 32 kHz, with 10 ms plateau, 1 ms cos2 rise/fall) or clicks of 0.03 ms were presented at 40 Hz (tone bursts) or 20 Hz (clicks) in the free field ipsilaterally using a JBL 2402 speaker.

### Patch-clamp recordings from IHCs

The apical turn of the organs of Corti were acutely dissected from P14 to P18 animals in HEPES Hank’s solution containing (in mM): 5.36 KCl, 141.7 NaCl, 10 HEPES, 0.5 MgSO4·7H2O, 1 MgCl2, 6H2O, 5.6 D-glucose, and 3.4 L-glutamine (pH 7.2, ∼300 mOsm/l) and was introduced into the recording chamber via a perfusion system. The pipette solution contained (in mM): 110 NaCl, 35 TEA-Cl, 2.8 KCl, 1 MgCl2, 1 CsCl, 10 HEPES, 5 CaCl2, and 11.1 D-glucose (pH 7.2, ∼305 mOsm/l) and was introduced into the recording chamber via a perfusion system. The pipette solution contained (in mM): 130 Cs-glucanate, 10 TEA-Cl, 10-4 AP, 10 HEPES, 1 MgCl2, as well as 300 mg/ml amphotericin B (pH 7.17, ∼290 mOsm/l). The measurements were undertaken with EPC-10 amplifiers controlled by Patchmaster software (HEKA Elektronik, Germany). IHCs were held at −84 mV. All voltages were corrected for liquid junction potential offline (14 mV). Currents were leak corrected using a p/10 protocol (0.4 pA/psec). Tone bursts were presented at 40 Hz (tone bursts) or 20 Hz (clicks) in the free field ipsilaterally using a JBL 2402 speaker.

### Data analysis

#### Immunohistochemistry

The intensities of AP180, otoferlin and synapsin1/2 fluorescence signals along the longitudinal axis of IHCs (Fig. 1) were measured via line profiles using ImageJ software. Igor Pro 6.3 software was used to generate average intensity profiles. The distribution of AP180 in the cytosol and its membrane proximity was assessed from intensity line scans reaching from the basal plasma membrane towards the cytosol along the longitudinal axis of IHCs (Fig. S1C–C′) and from a high-intensity fluorescence cluster analysis (Fig. S1D–D′) performed using ImageJ software. To do this, middle planes of IHCs were selected and cell borders were defined via otoferlin co-staining. In the otoferlin channel, lines from the cell membrane towards the nucleus were drawn and fluorescence intensity was measured in the AP180 channel (pixel intensity values ranging from 0 to 255) to avoid bias. Igor Pro 6.3 software was used to generate average intensity profiles of the individual cells. Separated clusters of high AP180 immunofluorescence intensity were defined by application of an intensity threshold (the threshold was adjusted once for clusters in a stimulated example image and then used for all images) using ImageJ software. Within each IHC (as defined by otoferlin co-staining), the average size and mean intensity of all clusters were calculated using the ImageJ tool ‘analyze particles’. Only clusters with a size of ≥0.04 μm² were included because of the resolution limit of confocal microscopy (∼200–250 nm for the used fluorophores here). IHC ribbon synapses were manually counted in z-projections of confocal sections from RIBEYE/CtBP2 and Homer1 immunolabeled organs of Corti using ImageJ software. Otoferlin and Vglut3 levels of IHCs were semi-quantitatively assessed as immunofluorescence intensity values and analyzed using Imaris (Bitplane) and Matlab (Mathworks), as described previously (Strenzke et al., 2016). Intensities of each cell are normalized to the average intensity of the Wt group for otoferlin or for Vglut3.

#### qPCR

StepOne™ Real-Time PCR System software was used to analyze qPCR results. Relative quantification values were calculated by comparative Ct method. Hprt was used as a housekeeping gene.

#### Electron tomography

Only tomograms with a single ribbon synapse clearly anchored to the AZ were selected for quantification using the IMOD package 3dmod (http://bio3d.colorado.edu/imod/) and as described previously (Chakrabarti et al., 2018; Kroll et al., 2019). Small, clear vesicles were classified as synaptic vesicles (SVs) if they appeared round and if their outer diameter (defined by fitting a circle to the borders at the maximum projection of the respective SV) was smaller than 70 nm. SVs were categorized as belonging to one of three groups: (1) ribbon-associated SVs (RA-SVs) if they were located in the first row around the synaptic ribbon and with a maximum distance of 80 nm from the ribbon, (2) membrane-proximal SVs (MP-SVs) if the distance between SV and plasma membrane was less than 50 nm and if the distance between SV and presynaptic density was less than 100 nm, and (3) cytosolic SVs if the distance between SV and ribbon was less than 500 nm thereby excluding RA-SVs and MP-SVs (all criteria being valid at maximum projection of the respective SV). MP-SVs were further subdivided into four groups depending on their connection to the AZ plasma membrane as described in a previous study (Chakrabarti et al., 2018). SVs were counted as docked if the distance between SV outer membrane and AZ membrane was 0–2 nm. Single-tethered SVs were defined as all SVs with only one tether reaching from the SV to the AZ membrane. Multi-tethered SVs were defined as all SVs with more than one tether from the SV to the AZ membrane. SVs were counted as membrane anchored if the distance between SV and plasma membrane (thereby ignoring possible filamentous connections between SVs, as well as between SVs and presynaptic density or between SV and ribbon). Furthermore, all structures containing a clathrin coat were counted as coated structures if the minimal distance between organelle and ribbon was less than 500 nm. This quantification includes structures with a coat around the entire clathrin-coated SV (independent of its size), coated pits budding from the plasma membrane and endosome-like vacuoles (ELVs) with at least one budding coated pit (in this case, the ELV with pit was only counted once and not included in the additional ELV quantification). All membrane organelles were defined as ELVs and counted if their maximum diameter was larger than 70 nm and/or if they were tubular, and if the minimal distance between ELV and ribbon was less than 500 nm. The volume of each ELV was assessed from a modeled mesh calculated from contours drawn in every virtual section of the ELV in 3dmod.

#### Patch-clamp recordings

Electrophysiological data was analyzed with custom-written programs in Igor Pro 6.3 and Python (available upon request). For the analysis of the I–V curves, the final 5 ms of the evoked Ca2+ currents were averaged after subtracting the mean of the first 5 ms of the recording. For the analysis of the stimuli train, 70 ms of Cm was averaged to calculate excitatory Cm increments.
\((C_{m\text{r}})\) skipping the first 50 ms after each depolarization to avoid impact of non-exocytic \(C_{m}\) transients. In order to estimate the replenishment rate of SVs, we undertook ‘SMN analysis’ as described previously (Schneguenburger et al., 1999). We plotted cumulative \(\Delta C_{m}\) versus the stimulus number. A line was fitted to the last five points, and the slope of the fit was taken as the replenishment rate. We furthermore calculated the replenishment rate in terms of vesicles per stimulus. We used the diameter values obtained from the tomograms of Wt and AP180-KO IHCs, subtracting the mean membrane thickness (Neef et al., 2007) to obtain the mid-membrane diameter. For the conversion of SV diameter to capacitance values, a specific capacitance value of 9.1 fF\(\mu\text{m}^{-2}\) (Albillos et al., 1997) was used and spherical shape was assumed.

For calculating the \(\Delta C_{m}\) the difference between the mean 400 ms \(C_{m}\) before and 200 ms after the depolarization was taken. The \(C^{2+}\) charge \((Q_{c})\) was calculated by taking the integral of the leak-subtracted current during depolarization. The endocytosis recordings were analyzed as described previously (Neef et al., 2014). The overall observed decrease in \(C_{m}\) during perforated patch-clamp recordings were corrected by using the slope of the global fit to \(C_{m}\) values taken from each recording. We calculated the slow component of the endocytosis by a linear fit of post-depolarization \(C_{m}\), skipping the first 200 ms. Recordings that did not return to baseline within 80 s were excluded from the analysis. The amplitude of the exponential subtraction of linear fit to final 15 s of \(C_{m}\). An exponential function was fit to \(C_{m}\) versus the

Sample analysis

Results

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Supplementary information

Supplementary information available online at http://jcs.biologists.org/lookup/doi/10.1242/jcs.236737.supplemental.

References

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Competing interests

The authors declare no competing or financial interests.

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