Altered dendritic spine function and integration in a mouse model of fragile X syndrome

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Cellular and circuit hyperexcitability are core features of fragile X syndrome and related autism spectrum disorder models. However, the cellular and synaptic bases of this hyperexcitability have proved elusive. We report in a mouse model of fragile X syndrome, glutamate uncaging onto individual dendritic spines yields stronger single-spine excitation than wild-type, with more silent spines. Furthermore, fewer spines are required to trigger an action potential with near-simultaneous uncaging at multiple spines. This is, in part, from increased dendritic gain due to increased intrinsic excitability, resulting from reduced hyperpolarization-activated currents, and increased NMDA receptor signaling. Using super-resolution microscopy we detect no change in dendritic spine morphology, indicating no structure-function relationship at this age. However, ultrastructural analysis shows a 3-fold increase in multiply-innervated spines, accounting for the increased single-spine glutamate currents. Thus, loss of FMRP causes abnormal synaptogenesis, leading to large numbers of poly-synaptic spines despite normal spine morphology, thus explaining the synaptic perturbations underlying circuit hyperexcitability.
Cell and circuit hyperexcitability have long been hypothesized to underlie many core symptoms of fragile X syndrome (FXS) and autism spectrum disorders more generally, which include sensory hypersensitivity, seizures and irritability. The fundamental role of cellular excitability in circuit function raises the possibility that alterations in neuronal intrinsic physiology may underlie a range of functional endophenotypes in FXS. Despite this potential link, few studies have examined the combined synaptic, dendritic, and cellular mechanisms that lead to generation of neuronal hyperexcitability during early postnatal development.

Many cellular properties are known to regulate neuronal excitability, such as neuronal morphology, intrinsic physiology, synaptic transmission and plasticity. In FXS, a central hypothesis is that glutamatergic signalling at dendritic spines is impaired, concomitant with changes to intrinsic cellular excitability. The first major alteration described was a change in dendritic spine density and morphology. However, this observation was not apparent when examined at the nanoscale using super-resolution imaging methods, despite an increase in synapse and spine density in the neocortex. Notwithstanding, no study has yet observed a change in synaptic event frequency that would be predicted by a change in spine or synapse density. This has important implications for our understanding of the synaptic aetiology of FXS, as many of the current theories are reliant on altered synaptic function.

The rodent somatosensory cortex (S1) is well characterised in terms of its processing of tactile inputs, which, in the case of the barrel cortex arise from the whiskers on the facepad via relay synapses in the brainstem and ventrobasal thalamus. The thalamic inputs arrive predominantly onto layer 4 stellate cells (L4 SCs) which integrate this information within L4, then project to L2/3 and L6. Furthermore, L4 SCs undergo a well-described critical period for synaptic plasticity, which closes at postnatal day 7–8. For these reasons, L4 of S1 provides a well-described reductionist system to examine sensory processing. Indeed, hyperexcitability has been observed within L1 of Fmr1 knockout mice, due in part to changes in intrinsic neuronal excitability, axonal morphology, and synaptic connectivity, which together result in increased network excitability. The finding that the critical period for thalamocortical synaptic plasticity is delayed in Fmr1 knockout mice compared with wild type (WT) gave a suggestion as to how cellular and circuit deficits may arise. How this delay in synapse development delay affects dendritic spine function is not known. Furthermore, no study has directly examined how dendrites integrate synaptic inputs in the absence of FMRP, despite the fact that dendritic integration plays a key role in regulating cellular excitability. Of particular relevance are findings that HCN channel expression is altered, leading to changes in intrinsic physiology and dendritic integration. Here, we directly test whether there is a functional relationship between dendritic spine function, intrinsic neuronal physiology, HCN channel function, dendritic integration, and ultimately neuronal output. To address this question, we use an integrative approach that combines whole-cell patch-clamp recording from neurons in S1 at P10–14 with 2-photon glutamate uncaging, post hoc stimulated emission-depletion (STED) microscopy, and serial block-face scanning-electron microscopy.

Results

Larger single dendritic spine currents in Fmr1−/− L4 SCs. To first assess the function of identified dendritic spines in Fmr1−/− mice, we performed single-spine 2-photon glutamate uncaging. Whole-cell patch-clamp recordings were performed from L4 SCs in voltage clamp with a Cs-glucuronate based intracellular solution containing a fluorescent dye (Alexafluor488, 100 μM) and bicuculline to allow on-line and post hoc visualisation of dendritic spines. Following filling, we performed 2-photon uncaging of Rubi-glutamate (Rubi-Glu) to elicit uncaging excitatory post-synaptic currents (uEPSCs; Fig. 1a). From both the concentration- and power–response relationships (Supplementary Fig. 1A, B), we determined that 300 μM [Rubi-Glu] and 80–100 mW laser power (λ/λ0 nm) were optimal to produce saturating uEPSCs at −70 mV. Analysis of the spatial properties of Rubi-Glu uncaging confirmed that the optimal position for photolysis was 0–1 μm from the edge of the spine head (Supplementary Fig. 1C), and the resulting uEPSCs were blocked with CNQX, confirming that they were produced by AMPA receptors (AMPARs, Supplementary Fig. 1D). We also found no difference in spine distance from cell soma and uEPSC rise or decay time and amplitude suggesting equal space clamp of the neurons across the dendritic distances examined (Supplementary Fig. 1F–H). All details of statistical tests performed can be found in Supplementary Table 1.

Comparison between genotypes revealed that the single-spine uEPSCs in WT mice had an amplitude of 6.9 ± 0.4 pA (n = 17 mice), while Fmr1−/− mice (n = 14 mice) showed a larger uEPSC amplitude of 9.8 ± 0.5 pA (d.f.: 4, 5; χ² = 8.26; p = 0.004; LMM, Fig. 1 and Supplementary Fig. 2), indicating that spines in Fmr1−/− mice are enriched for AMPAR-mediated currents (Fig. 1b, c). This difference appeared to be due to a larger population of uEPSCs at Fmr1−/− spines with amplitudes over 10 pA (Fig. 1b). As expected from larger underlying currents, the single-spine uncaging excitatory post-synaptic potential (uEPSP) was also larger in Fmr1−/− mice (0.73 ± 0.12 mV, n = 10 mice), when compared with WT littermates (0.47 ± 0.06 mV, n = 16 mice; d.f.: 24; t = 2.09; p = 0.046; T-test; Fig. 1d). In a subset of dendritic spines we observed no AMPAR current at −70 mV, however, a large NMDA receptor (NMDAR) current was present at +40 mV, indicating the presence of silent dendritic spines (Fig. 1e). Quantification of the silent spines revealed an occurrence of 17.6 ± 3.5% in Fmr1−/− mice (n = 13 mice), almost threefold higher than in WT mice (6.4 ± 1.6%, n = 17 mice; d.f.: 27; t = 3.1; p = 0.005; T-test; Fig. 1f). When measured across all spines, the NMDA/AMPA ratio was significantly elevated as both a population average (d.f.: 1, 331; F = 37.36; p < 0.0001; F-test; Fig. 1g) and also as a spine average with Fmr1−/− mice having a ratio of 1.26 ± 0.05 (n = 117 spines) and WT of 0.97 ± 0.03 (n = 194 spines; χ² = 6.27 p = 0.012, LMM, Fig. 1h and Supplementary Fig. 3).

Given that the majority of L4 SC dendritic spines are formed by cortico-cortical synapses in WT mice, and therefore likely comprise the majority of uncaged spines, we next asked whether synapses formed between L4 SCs had larger EPSC amplitudes by performing paired recordings between synaptically coupled neurons (Fig. 2). As previously described in 2-week-old mice, we observed a low connectivity between L4 SCs in Fmr1−/− mice of 14.8%, that is significantly lower than that of WT mice which had a connectivity of 33.6% (p = 0.015, Fisher’s exact test, Fig. 2c). Despite this reduced connectivity, there was no difference in either failure rate (d.f.: 41; t = 0.25, p = 0.80; GLMM; Fig. 2d) or unitary EPSC amplitude (d.f.: 41; t = 1.53, p = 0.15; LMM; Fig. 2e), suggesting that synaptic strength is unchanged at the majority of synapses in Fmr1−/− mice.

Fmr1−/− spines have typical morphology but more synapses. The inclusion of bicuculline within the internal solution allowed post hoc visualisation of the recorded neurons, following fixation and re-sectioning. We next performed correlated stimulated emission-depletion (STED) imaging of the same dendritic spines we had uncaged upon (Fig. 3a–e). Measurement of nanoscale spine...
**Fig. 1** L4 SC dendritic spines have larger uEPSCs with more silent synapses in Fmr1−/y mice.  

**a** 2-photon image of a L4 SC (left) with selected spines and AMPAR uEPSCs from WT and Fmr1−/y mice. Scale bars: 20 µm (left), 5 µm (right).  

**b** Single-spine uEPSCs from WT (black) and Fmr1−/y (red) mice shown as a histogram, with spine average shown (inset). Note that spines with no AMPA response, silent spines have not been included.  

**c** Animal average uEPSC amplitudes, excluding silent spines. Number of animals tested shown in parenthesis.  

**d** Animal average of uEPSP amplitudes.  

**e** AMPAR (upper) and NMDAR (lower) uEPSCs, illustrating silent spines. Scale: 5 µm.  

**f** Incidence of silent spines in WT and Fmr1−/y mice.  

**g** AMPAR and NMDAR uEPSCs for all spines, with NMDA/AMPA ratio (WT: 0.76 ± 0.03; Fmr1−/y: 1.05 ± 0.04; d.f.: 1, 331; F = 37.4; p < 0.0001; F-test).  

**h** Average NMDA/AMPA ratio plotted for all spines. Statistics shown: *p < 0.05, **p < 0.01, from LMM (b, d, h), unpaired t-test (c, f) and sum-of-least-squares F-test (g). Plots of individual spine data for panels c (inset) and h can be found in Supplementary Fig. 4. All data are shown as mean ± SEM and source data for all plots are provided as a Source Data file.
morphology revealed that there was no difference in either spine head width (Fig. 3b), nor neck length (Fig. 3d), between WT (n = 6 mice) and Fmr1−/− (n = 4 mice) mice. Consistent with earlier findings, we observed a weak positive correlation with spine head width and EPSC amplitude in WT mice (7.8 ± 3.8 pA/µm, R² = 0.06, F = 4.3, p = 0.042, F-test), which was not different to that of Fmr1−/− mice (F = 0.02, p = 0.89, sum-of-squares F-test; Fig. 3c). We observed no correlation with spine neck length and EPSC amplitude (Fig. 3e). To confirm that uncaging itself did not result in spine remodelling, we also measured spines from non-uncaged dendrites on filled neurons. Spine density itself was not different between genotypes (Fig. 3i, j), in agreement with previous findings from L5 of S1 and CA1 of the hippocampus.

Given the strengthening of dendritic spines, but no change in unitary EPSC amplitude or spine morphology, we next asked whether the ultrastructure of dendritic spines was altered. To achieve this, we used serial block-face scanning-electron microscopy in L4 of S1 from mice perfusion fixed at P14. In serial stacks (50 nm sections; Fig. 4) we identified Type-1 asymmetric synapses on dendritic spines, based on the presence an electron dense post-synaptic density (PSD) opposing an axon bouton containing round vesicles. Following 3-dimensional reconstruction, we identified a subset of dendritic spines that contained more than one PSD, which were each contacted by an independent presynaptic axon bouton (Fig. 4a, b), and henceforth referred to as multi-innervated spines (MIS). These MIS were present in both genotypes, however, the incidence in Fmr1−/− mice was 20.5 ± 1.6% of all spines (n = 7 mice), approximately threefold higher than in WT littermates (7.2 ± 1.5% of spines, n = 3 mice, d.f.: 8; t = 4.9; p = 0.001; T-test; Fig. 4c), which is similar to that observed in organotypic hippocampal cultures from WT mice.

The presence of higher numbers of MIS in Fmr1−/− mice, and larger single spines uEPSCs, despite a similar density of spines and similar dendritic morphologies, would suggest an increased number of synapses for each L4 SC. The conventional method to assess such a change in synapse number is to perform miniature EPSC (mEPSC) recordings (Fig. 5a). AMPAR mEPSCs recorded at −70 mV in Fmr1−/− mice were very similar to WT in both amplitude (d.f.: 46; U = 245; p = 0.28; Mann–Whitney test) and frequency (d.f.: 46; U = 240; p = 0.24; Mann–Whitney test; Fig. 5b). NMDAR mEPSCs, recorded at +40 mV in the presence of CNQX, also had very similar amplitudes (d.f.: 17; U = 37; p = 0.59; Mann–Whitney test). However, Fmr1−/− mice showed a 54% increase in NMDAR mEPSC frequency compared with WT mice (d.f.: 17; U = 18; p = 0.03; Mann–Whitney test; Fig. 5c). These data indicate that while AMPAR-containing synapses number and strength are unaltered in Fmr1−/− mice, they possess ~50% more NMDAR containing synapses.

**Fmr1−/− L4 SCs are hyperexcitable due to lower HCN currents.** While these observed changes in synaptic properties reveal differences in dendritic spine function, alone they do not reveal how neurons integrate excitatory inputs leading to hyperexcitability. Dendritic spines act as spatiotemporal filters whose summation is dependent upon synaptic receptor content and intrinsic membrane properties, the latter of which contributes to the cable properties of dendrites. To explore the effect of altered synaptic properties on dendritic integration in Fmr1−/− SCs, we next measured the intrinsic excitability of L4 SCs by assessing their

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**Fig. 2** Typical EPSC amplitude at unitary connections between L4 SCs. a Schematic paired recordings between synaptically coupled L4 SCs. b Representative presynaptic action potentials (top) produced unitary EPSCs in the second L4 SC (lower), from WT (black) and Fmr1−/− (red) mice. c Synaptic connectivity is reduced between L4 SCs in the Fmr1−/− mouse (d.f.: 162; p = 0.015; Fisher’s exact test; 110 pairs from 13 mice for WT mice and 54 pairs from 7 mice in Fmr1−/− mice were tested. d Failure rate was not different between genotypes when a connection was present. e Unitary EPSC amplitudes from L4 SC synapses were not different between genotypes. Statistics shown: ns

**Fig. 3** a Connectivity in WT and Fmr1−/− mice (110 −/− SCs, 110 WT SCs). b Head width and neck length in WT and Fmr1−/− mice. c Connectivity (%), d Head width and e Neck length (%). All graphs show mean ± SEM and source data for all plots are provided as a Source Data file.
Fig. 3 Dendritic spines show no difference in nanoscale morphology, or structure-function relationship. a) Dendrites from WT (left) and Fmr1<sup>−/−</sup> (right) mice under 2-photon microscopy (top), then post hoc STED imaging (bottom). Scale bar: 5 μm. b) Average spine head width in WT (black) and Fmr1<sup>−/−</sup> (red) mice (WT: 0.43 ± 0.05; Fmr1<sup>−/−</sup>: 0.49 ± 0.04; d.f.: 8; t = 0.29; p = 0.78, T-test). Number of mice is indicated. c) Comparison of spine head width and uEPSC amplitude (comparing slope: d.f.: 1; 100; F = 0.62; p = 0.89). WT spines showed a positive correlation (d.f.: 70, F = 4.27, p = 0.042, F-test). d) Average spine neck length (WT: 1.52 ± 0.22; Fmr1<sup>−/−</sup>: 1.31 ± 0.20; d.f.: 8; t = 0.66; p = 0.53, T-test). e) Comparison of spine neck-width and uEPSC amplitude (Slope: WT: 2.1 ± 0.8; Fmr1<sup>−/−</sup>: 0.8 ± 1.4; d.f.: 1; 101; F = 0.84; p = 0.36, F-test). f) Spine density on L4 SCs (WT: 6.8 ± 0.7 spines/10 μm; Fmr1<sup>−/−</sup>: 6.1 ± 0.80 spines/10 μm; d.f.: 13; t = 0.60; p = 0.56; T-test). g) Distribution of non-uncaged spine head-widths, as an average of all mice (bold) and individual mice (dashed). h) Average head width of non-uncaged spines (WT: 0.48 ± 0.05 μm; Fmr1<sup>−/−</sup>: 0.48 ± 0.04 μm; d.f.: 13; U = 20.0; p = 0.59; Mann-Whitney U-test). i) Distribution of spine neck length of non-uncaged spines. j) Average spine neck length in non-uncaged spines (WT: 1.36 ± 0.12 μm; Fmr1<sup>−/−</sup>: 1.27 ± 0.14 μm; d.f.: 13; U = 20.0; p = 0.55; Mann-Whitney U-test). Statistics shown: ns > 0.05 from unpaired t-test (b, d, f, h, j) and sum-of-least-squares F-test (c, e). All data are shown as mean ± SEM and source data for all plots are provided as a Source Data file.

response to hyperpolarising and depolarising current injections (Fig. 6a, b). In Fmr1<sup>−/−</sup> mice, L4 SC input resistance (R<sub>L</sub>) was increased compared with WT mice, as measured from the steady-state current–voltage relationship (Interaction: d.f.: 5, 230; F = 7.03; p < 0.0001; two-way RM ANOVA Fig. 6c) and smallest current step response (d.f.: 222; t = 2.21, p = 0.023; GLMM; Fig. 6c, inset). This increase in R<sub>L</sub> in Fmr1<sup>−/−</sup> mice was associated with an increase in action potential (AP) discharge (Interaction: d.f.: 5, 230; F = 6.17; p < 0.0002; two-way RM ANOVA, Fig. 6d), resulting from a decreased rheobase currents in the recorded L4 SCs (d.f.: 222; t = 2.15, p = 0.035; GLMM, Fig. 6d, inset). The dynamic response of neurons to modulating current when measured with a sinusoidal wave of current injection (0.2–20 Hz, 50 pA, 20 s duration, Fig. 6e) led to a resonant frequency of 1.1 ± 0.1 Hz in L4 SCs from Fmr1<sup>−/−</sup> mice, which was higher than that of 0.8 ± 0.1 Hz in WT littermates (d.f.: 25; t = 3.25; p = 0.002; LMM; Fig. 6f). Furthermore, there was no change in resonant dampening (Q-factor: WT: 1.23 ± 0.07; Fmr1<sup>−/−</sup>: 1.13 ± 0.03; d.f.: 24; t = 0.7; p = 0.49; T-test) indicating equally sustained activity at these frequencies between genotypes. Further analysis of passive membrane properties (Supplementary Fig. 6b and C) did not reveal genotype-specific differences. While AP amplitude was minimally reduced (Supplementary Fig. 6e), no other parameter was significantly altered, confirming the specificity of R<sub>L</sub> leading to altered cellular excitability. These analyses demonstrate that L4 SCs from Fmr1<sup>−/−</sup> mice are intrinsically more excitable than their WT counterparts.

In S1 L5 pyramidal cells, HCN channel density is reduced leading to reduced I<sub>h</sub> as measured indirectly as a voltage sag in current-clamp<sup>17,22</sup>. Therefore, we next asked whether I<sub>h</sub> mediated sag is also reduced in L4 SCs and contributes to the genotypic differences in intrinsic excitability we have observed. We first measured the sag and membrane rebound in response to hyperpolarising current steps in current-clamp from −60 mV (0 to −125 pA, 25 pA steps, 500 ms duration; Fig. 7a). The voltage sag, as measured as a percentage of the maximum hyperpolarisation (Fig. 7b) was significantly reduced in Fmr1<sup>−/−</sup> mice (7.6 ± 0.6% of maximum) when compared with WT controls (10.9 ± 0.5% of maximum, d.f.: 218; t = 3.59; p = 0.0003; GLMM), indicating reduced I<sub>h</sub>. A further measure of I<sub>h</sub> is the rebound...
Potential produced on return to \(-60\) mV\(^2\). Consistent with reduced srg, we observed a lower rebound potential in \(Fmr1^{-/-}\) L4 SCs when measured relative to the steady-state potential (Fig. 7c). Furthermore, the rebound slope from individual cells was \(-0.09 \pm 0.01\) mV/mV in \(Fmr1^{-/-}\) neurons, lower than that of WT (\(-0.11 \pm 0.01\) mV/mV, d.f.: 207; \(t = 2.28, p = 0.024\); LMM, Fig. 7d).

We next applied the \(I_h\) blocker ZD-7288 (ZD; 20 \(\mu\)M) to a subset of cells to assess the effect of \(I_h\) on intrinsic excitability. We observed a tendency to greater \(R_i\) in \(Fmr1^{-/-}\) than in WT mice (d.f.: 57; \(t = 1.85, p = 0.078\); LMM, Fig. 7e), similar to that we had observed previously (Fig. 6c). Following ZD application in WT L4 SCs, \(R_i\) increased by 49\% (d.f.: 28; \(t = 6.05, p = 1.99 \times 10^{-7}\); LMM; Fig. 7e), while \(Fmr1^{-/-}\) L4 SCs only showed a 14\% increase (d.f.: 28; \(t = 1.28, p = 0.20\); LMM; Fig. 7e). The \(ZD\) effect on \(R_i\) was significantly lower \(Fmr1^{-/-}\) L4 SCs compared with WT (d.f.: 57; \(t = 4.37, p = 6.3 \times 10^{-5}\); LMM; Fig. 7f). Given the observed differences in AP discharge between genotypes (Fig. 6d), we next tested whether ZD normalised this genotypic difference. In WT L4 SCs, ZD application significantly increased AP firing (d.f.: 5, 80; \(F = 3.2\); \(p = 0.011\) for interaction; two-way RM ANOVA; Fig. 7g). However, ZD had no effect on the AP discharge of \(Fmr1^{-/-}\) L4 SCs (d.f.: 5, 174; \(F = 0.23\); \(p = 0.95\) for interaction; two-way ANOVA; Fig. 7h), consistent with reduced sag. Finally, we examined the effect ZD had on the resonance of L4 SCs. In WT L4 SCs, ZD increased the impedance at low frequencies by 33\% (d.f.: 15; \(t = 2.66, p = 0.017\); GLMM; Fig. 7i, k), whereas ZD had no effect on impedance in \(Fmr1^{-/-}\) neurons (d.f.: 13; \(t = 0.83, p = 0.41\); GLMM; Fig. 7j, k). These data show that the intrinsic excitability of L4 SCs is increased in \(Fmr1^{-/-}\) mice, with WT L4 SC excitability increased by ZD application, potentially explaining genotype-specific differences in cellular intrinsic excitability.

Voltage sag and rebound are indicative of altered \(I_h\). To directly measure \(I_h\) in L4 SCs we next performed dedicated voltage-clamp experiments using a paradigm described previously\(^9\). \(I_h\) was recorded from L4 SCs in the presence of sodium channel, potassium channel, calcium channel, and GABA\(_\alpha\) receptor blockers, as well as AMPA and NMDA antagonists, from –50 mV with hyperpolarising steps (10 mV steps, 5 second duration, Fig. 8a). \(I_h\) had a half-maximal activation potential \((V_{1/2,\text{max}})\) in WT L4 SCs of \(–86\) mV, which in \(Fmr1^{-/-}\) was more hyperpolarised at \(–92\) mV (d.f.: 4, 584; \(F = 4.58, p = 0.001\); F-test; Fig. 8b). Despite this difference, \(I_h\) elicited at the most hyperpolarised voltage steps was similar (d.f.: 1, 370; \(F = 0.001, p = 0.97\); F-test), suggesting a normal complement of HCN channels (these currents in both WT and \(Fmr1^{-/-}\) L4 SCs were sensitive to ZD, Fig. 8b, inset). As the activation of \(I_h\) is directly associated to the intracellular cyclic-AMP concentration\(^8\), we next asked if increasing intracellular cyclic-AMP could rescue \(I_h\) activation in \(Fmr1^{-/-}\) neurons. To increase cyclic-AMP levels, we bath applied the adenylyl cyclase activator forskolin (50 \(\mu\)M) to the bath. Forskolin significantly increased the activation of \(I_h\) in both WT and \(Fmr1^{-/-}\) L4 SCs (Fig. 8c), normalising the \(I_h\) activation curves between genotypes (d.f.: 4, 310; \(F = 0.2, p = 0.94\); F-test, Fig. 8d). This data indicates that the decrease in \(I_h\) and hence increase in intrinsic excitability, in \(Fmr1^{-/-}\) L4 SCs results from a reduced cAMP-mediated shift in HCN activation.

**Enhanced dendritic summation in L4 SCs from \(Fmr1^{-/-}\) mice.**

Given that NMDARs and HCN channels are key determinants of dendritic integration\(^19,20\), we next assessed both spatial and temporal dendritic summation in the \(Fmr1^{-/-}\) L4 SCs. To address spatial summation in L4 SC dendrites we performed near-simultaneous glutamate uncaging at multiple spines (Fig. 9a), by focal puff application of Rubi-Glu (10 \(\mu\)M) and rapidly uncaged on dendritic spines (0.5 ms/spine). We first performed a sequential uncaging (i.e. each spine individually), then near-simultaneous uncaging of spine ensembles (i.e. groups of spines; Fig. 9b).

Summatng EPSPs ultimately resulted in a AP discharge from L4 SCs. \(Fmr1^{-/-}\) L4 SCs required activation of fewer spines on average to initiate an AP (d.f.: 23; \(t = 2.3\); \(p = 0.03\); T-test; Fig. 9c), which was more pronounced when silent spines excluded from analysis (d.f.: 18; \(t = 3.2\); \(p = 0.005\)). In five \(Fmr1^{-/-}\) L4 SCs, uncaging at spines individual was not performed, thus were not included in further analysis. Measurement of the summated EPSP, with respect to number of spines near-simultaneously uncaged showed that both WT and \(Fmr1^{-/-}\) L4 SC dendrites showed an increase in EPSP amplitude with increasing number of spines (Fig. 9d), which was significantly greater in the \(Fmr1^{-/-}\) L4 SCs (d.f.: 1, 170; \(F = 8.98, p = 0.003\); F-test). This measure will include effects due to increased spine synaptic strength and input resistance, in addition to dendritic integrative properties. Therefore, we next compared the expected linear sum of single-spine EPSPs with that of the observed summated EPSP (Fig. 9e), thereby excluding individual spine strength and input resistance effects on EPSP amplitude. We observed sublinear integration in WT and \(Fmr1^{-/-}\) L4 SCs, however, WT neurons showed low levels of integration (Slope: \(0.50 \pm 0.09\)), while \(Fmr1^{-/-}\) neurons presented over 50\% higher summation (Slope: 0.79 \pm 0.08; d.f.: 1, 195; \(F = 3.18, p = 0.044\); F-test). These data clearly show that the dendrites of \(Fmr1^{-/-}\) L4 SCs undergo excessive dendritic summation of synaptic inputs. To confirm that dendritic
summation is altered in response to endogenous synaptic transmission, we next provided extracellular stimulation to thalamocortical afferents (TCA) from the ventrobasal thalamus, whilst recording from L4 SCs (Fig. 9f). Stimulus intensity was titrated so that an EPSC of ~150 pA was produced, then trains of stimulation 55 ± 13% of the time (d.f.: 16; t = 3.81; p < 0.01; F-test; Supplementary Fig. 6D) and also prolonged decay times of the first EPSP (Fig. 9f, d.f.: 15; t = 2.34; p = 0.034; T-test; Supplementary Fig. 9C). ZD had no observable effect on summating EPSPs in Fmr1−/− L4 SCs (Supplementary Fig. 9E). Finally to confirm that altered Ih and NMDAR function contribute to the observed aberrant dendritic summation, in a subset of experiments we examined the effects of both ZD and AP-5 on EPSP summation during multispine uncaging. Application of either ZD or AP-5 to near-simultaneous uncaging of uEPSPs in WT L4 SCs had minimal effect on the observed summation when compared with the expected linear sum (Supplementary Fig. 10A), consistent with an absence of non-linear summation. However, both application of either ZD or AP-5 significantly reduced the summation of Fmr1−/− L4 SCs (Supplementary Fig. 10B). These findings confirm that both reduced HCN activation and increased NMDARs contribute to the enhanced summation in dendrites of Fmr1−/− L4 SCs relative to WT cells.

Discussion

L4 of the primary somatosensory cortex is the first layer to receive and integrate incoming sensory information, which is integrated and relayed within the cortex. As such, L4 SCs play a crucial role in sensory perception. Individuals with FXS show altered sensory processing, and mouse models show altered circuit processing in primary sensory areas. Furthermore, while FMRP has been shown repeatedly to regulate synapse function and plasticity, little is known about how these alterations affect dendritic spine function and dendritic integration to sensory input. To address these questions, we used glutamate uncaging at L4 SC dendritic spines to examine how they integrate and generate action potentials following synaptic stimulation. We show that L4 SCs in S1 have dendritic and synaptic properties that result in increased action potential generation in Fmr1−/− mice relative to WT controls. Specifically, we show increased excitatory synaptic currents at individual spines resulting from increased AMPAR and NMDAR content. Despite this, we observed no change in spine morphology using STED microscopy.
and there was little correlation between spine structure and function, indicating that spine morphology is not an effective proxy for spine function, at least at the age used in this study. However, electron microscopic analysis revealed an increase in multiply innervated spines which likely accounts for the increase in single-spine synaptic currents. Interestingly, there was also an increase in silent spines which agrees with the increase in NMDAR mEPSC frequency, but not AMPAR mEPSC frequency.

The overall increase in dendritic spine currents was accompanied by enhanced dendritic integration likely resulting, at least in part, from a ~50% reduction in \( R_i \). This reduced \( R_i \) was causal to the altered intrinsic physiology of L4 SCs at P12–14. Finally, TCA stimulation at frequencies that fail to elicit AP discharge from L4 SCs in WT mice, in the presence of intact synaptic inhibition, reliably elicits APs in \( Fmr1^{-/-} \) neurons, indicating that the local inhibitory circuit cannot compensate for the increase in synaptic and dendritic excitability. Together these findings demonstrate that aberrant dendritic spine function and dendritic integration combine to result in cellular hyperexcitability in L4 SCs. As the first cortical cells to receive input from the sensory periphery, the resultant hyperexcitability likely contributes previously reported circuit excitability in \( Fmr1^{-/-} \) mice and the sensory hypersensitivities in individuals with FXS.

Our study quantifies the incidence of MIS in intact tissue and implicates their presence in pathological states associated with disease models. Indeed, the mean increase in spine uEPSC amplitude, but not miniature, spontaneous or unitary EPSCs, in \( Fmr1^{-/-} \) mice is likely caused by the increase in the number of MIS. Indeed, the presence of MIS in both WT and \( Fmr1^{-/-} \) mice disagrees with the one spine/one synapse hypothesis. A potential mechanistic link between loss of FMRP and the increase in MIS may come from its ability to regulate PSD-95. PSD-95 mRNA is a known FMRP target and an increase in PSD-95 puncta in L4 of S1 has been observed with no change in cell number, dendritic morphology, or spine density in \( Fmr1^{-/-} \) mice. Furthermore, transient overexpression of PSD-95 results in increased MIS incidence through nitric oxide synthase, as well as NMDARs and other LTP mechanisms. Future experiments exploring the effect of NOS blockade, PSD-95, and NMDAR function in \( Fmr1^{-/-} \) mice should test the mechanism of
Fig. 7  

**a** Hyperpolarizing steps in L4 SCs (0 to −125 pA, 25 pA steps, 500 ms duration) with voltage “sag” and rebound potential indicated, as measured in WT (black, left) and *Fmr1*<sup>−/−</sup> mice (red, right). 

**b** Quantification of voltage sag expressed as % of maximum voltage for WT and *Fmr1*<sup>−/−</sup> L4 SCs. 

**c** Plot of rebound potential as a function of steady-state voltage for WT and *Fmr1*<sup>−/−</sup> L4 SCs, fitted with linear regression and with fit values displayed. 

**d** Quantification of the rebound slope of individual L4 SCs for both genotypes. 

**e** Change in input resistance before and after bath application of the *I<sub>h</sub>* blocker ZD-7288 (ZD; 20 µM) in WT and *Fmr1*<sup>−/−</sup> L4 SCs. 

**f** Change in input resistance (%) of ctrl before and after bath application of ZD in WT and *Fmr1*<sup>−/−</sup> L4 SCs. 

**g** Current–frequency plot of AP discharge before (solid lines) and after (dashed lines) ZD application. 

**h** The same analysis as in (g), but in *Fmr1*<sup>−/−</sup> L4 SCs. 

**i** Subthreshold membrane chirps (0.2–20 Hz, 50 pA, 20 s duration) and current–impedance plot for WT L4 SCs before (black) and after (grey) ZD application. 

**j** The same data as in (f), but in *Fmr1*<sup>−/−</sup> mice. 

**k** Impedance measured at peak resonant frequency in WT and *Fmr1*<sup>−/−</sup> L4 SCs before and after ZD (+ZD) application. 

Statistics shown: ns p > 0.05, *p < 0.05, **p < 0.01, ***p < 0.001, from LMM (b, d, e, f, k). Summary plots of all data shown in (b) and (d) can be found in Supplementary Fig. 7. All data are shown as mean ± SEM and source data for all plots are provided as a Source Data file.
MIS formation and influence on dendritic protein synthesis, as well as potential therapeutic targeting.

Interestingly, the increase in spines with increased uEPSC amplitudes and MIS was mirrored by an increase in silent TCA synapses, though their number was insufficient to compensate for the overall increase in dendritic currents in other spines. An increase in silent TCA synapses at P718 was previously reported for the overall increase in dendritic currents in other spines. While dendritic spines are functionally disrupted in the Fmr1−/y mouse, there is no indication of altered spine morphology or synaptic function in these neurons. This is in good agreement with previous findings that spine morphology is unaffected in hippocampal CA1 and layer 5 S1 neurons.

While dendritic spines are functionally disrupted in the Fmr1−/y mouse, using super-resolution microscopy we found no evidence of a genotypic difference in spine morphology of L4 SC neurons. This is in good agreement with previous findings that spine morphology is unaffected in hippocampal CA1 and layer 5 S1 neurons. Furthermore, we find only a weak correlation between dendritic spine structure and function, demonstrating the pitfalls of using spine structure as a proxy for synaptic function, especially in young animals and genetic models of disease. These findings are in stark contrast to those observed from post-mortem human tissue or from other mouse studies; however, these studies were only performed with diffraction-limited microscopy, suggesting that super-resolution imaging techniques should be the gold-standard for dendritic spine morphological studies in future. Single dendritic spines do not typically produce AP discharge from neurons, rather they require co-activation and summation of multiple synaptic inputs arriving with high temporal precision. L4 SCs have been previously been shown to possess linear integration of Ca2+ influx in their dendrites. We show that synaptic potentials sublinearly integrate in L4 SCs of WT mice, and that this integration is strongly enhanced in Fmr1−/y mice, leading to more efficient discharge of APs, due in large part to a combination of increased NMDARs and reduced Ih. The latter has been implicated in the altered neuronal excitability of FXS, with the HCN1 channel expression dictating whether the current is increased or decreased. Unlike these former studies, we provide evidence that Ih is not reduced in L4 SCs, but rather displays shifted activation properties, likely due to reduced cyclic-AMP levels. This finding in in agreement with previous work implicating altered AMP levels in the aetiology of FXS. Whether the altered Ih currents in the absence of FMRP reported in other cell types could also be explained by altered AMP levels is not known; however, at least for layer 5 neurons in somatosensory cortex, a reduced level of HCN channels has also been reported. Future experiments will be needed to determine the developmental and cell-specific nature of cellular hyperexcitability in Fmr1−/y mice.

Our observations showing sublinear dendritic integration in layer 4 SCs are at odds with reported NMDAR-dependent non-linear (supra-linear) summation of cortical cells reported from many laboratories. However, many factors may account for this discrepancy, including recording conditions, stimulation conditions, etc.
paradigms, cell type and developmental age. Furthermore, the somatosensory cortex has a well-described developmental profile of membrane properties, notably decreasing membrane resistance as a function of age. This combined with the compact dendritic arbour of L4 SCs, will lead to these neurons at the age of ~14 days likely having very uniform cable properties. Irrespective of the differences between studies, it is possible that as L4 SCs mature, their dendrites may develop non-linear properties. Irrespective of the differences between studies, it is possible that as L4 SCs mature, their dendrites may develop non-linear properties.

**Methods**

**Animals and ethics.** All procedures were performed in line with Home Office (ASPA, 2013; H0 license: P1351480E) and institutional guidelines. All experiments were performed on C57/Bl6J mice, bred from $Fmr1^{-/-}$ mothers, cross-bred with $Fmr1^{+/-}$ male mice, giving a Mendelian 1:1 ratio of $Fmr1^{+/-}$ and $Fmr1^{-/-}$ amongst male offspring. Only male mice were used for the present study and all mice were killed at P10-15, before separation from the mother. Mothers were given ad libitum access to food and water and housed on a 12 h light/dark cycle. All experiments and analysis were performed blind to genotype.

**Acute slice preparation.** Acute brain slices were prepared similar to previously described. Briefly, mice were decapitated without anaesthesia and the brain rapidly removed and placed in ice-cold carbogenated (95% O2/5% CO2) sucrose-modified artificial cerebrospinal fluid (in mM: 7 NaCl, 2.5 KCl, 25 NaHCO3, 1.25 NaH2PO4, 25 glucose, 75 sucrose, 7 MgCl2, 0.5 CaCl2). 400 μm thick thalamo-cortical (TC) slices were then cut on a Vibratome (VT1200s, Leica, Germany) and then stored submerged in sucrose-ACSF warmed to 34 °C for 30 minutes and transferred to room temperature until needed.

**Whole-cell patch-clamp recordings.** For electrophysiological recordings slices were transferred to a submerged recording chamber perfused with carbogenated normal ACSF (in mM: 125 NaCl, 2.5 KCl, 25 NaHCO3, 1.25 NaH2PO4, 25 glucose, 1 MgCl2, 2 CaCl2) maintained at near physiological temperatures (32 ± 1 °C) with an inline heater (LinLab, Scientifica, UK) at a flow rate of 6–8 ml/min. Slices were visualized with IR-DIC illumination (BX-51, Olympus, Hamburg, Germany) initially with a x4 objective lens (N.A. 0.1) to position above a L4 barrel, and then with a x20 water-immersion objective (N.A. 1.0, Olympus). Whole-cell patch-clamp recordings were made with a Multiclamp 700B amplifier (Molecular Devices, USA). Recording pipettes were pulled from borosilicate glass capillaries.

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**Fig. 9** Enhanced dendritic integration of L4 SCs in $Fmr1^{-/-}$ mice. a Schema of near-simultaneous glutamate uncaging (Rubi-Glu) at multiple spines (blue dots/numbers). b Near-simultaneous glutamate uncaging produced subthreshold (inset, right) and suprathreshold uEPSPs (inset, left) along dendrites.

c The number of spines required to evoke an AP, from all spines (left; WT: 8.8 ± 0.7; $Fmr1^{-/-}$: 6.6 ± 0.6) and excluding “silent spines” (right; WT: 8.7 ± 0.7; $Fmr1^{-/-}$: 5.6 ± 0.7). d Summation of near-simultaneous subthreshold uEPSPs normalized to the first EPSP in WT (black) and $Fmr1^{-/-}$ (red) L4 SCs (Slope: WT: 1.1 ± 0.13; $Fmr1^{-/-}$: 1.9 ± 0.2; d.f.: 1, 170; F = 8.98; p = 0.003; F-test).

d Summating uEPSPs plotted against the expected linear sum. Unity is indicated (grey).

e Electrical stimulation of TCA at low frequency 10 Hz is shown. f Average spike probability in response to 5 and 10 Hz stimulation. Statistics shown: *p < 0.05, **p < 0.01. All data are shown as mean ± SEM and source data for all plots are provided as a Source Data file.
Sequenential dendritic spine 2-photon glutamate uncaging. Slices were transferred to the recording chamber, which was perfused with ACSF, containing 50 µM picrotoxin (PTX) and 300 nM tetrodotoxin (TTX). For voltage-clamp recordings of dendritic spine uncaging neurons were filled with an internal solution containing (in mM): 140 Na-gluconate, 3 CaCl₂, 0.5 EGTA, 10 HEPES, 2 Mg-ATP, 2 Na₂-ATP, 0.3 Na₂-GTP, 1 phosphocreatine, 5 QX-314 chloride, 0.1% biotinoylated-lysine (Bioquant, Invitrogen, UK), and 0.1 AlexaFluor 488 or 594 (Invitrogen, UK), corrected to pH 7.4 with CsOH, Osm = 295–305 mOs. Whole-cell patch-clamp was then achieved and cells allowed to dye fill for 10 min prior to imaging. During the timed period, 5 min of spontaneous recordings were used to analyse mEPSCs from recorded neurons at ~70 mV voltage clamp. For all imaging and uncaging experiments we used a galvanometric scanning 2-photon microscope (Femto2D-Galvo, Femtontics, Budapest, Hungary) fitted with a femtosecond aligned, tunable wavelength Ti:Sapphire laser (Chameleon, Coherent, CA, USA), controlled by a Pockels cell (Conoptics, CT, USA). Following dye filling, a short, low zoom × stack was collected (2 µm steps, 2–3 pixel averaging, 512 × 512 pixels) over the whole dendritic extent of the cell at low laser power (< 5 mW) with a high numerical aperture ×20 lens (N.A. 1.0, Olympus, Japan). Then a short section of spine dendrite, 50–100 µm from the cell somata, within the top 50 µm of the slice, and running parallel to the slice surface was selected and imaged at 3 pixel averaging. Between 7 and 10 spines were then selected based on being morphologically distinct from neighbouring spines, ordered distal to proximal to soma, and then ~300 µM Rubi-Glutamate (Rubi-Glu; Ascent Scientific, Bristol, UK) was applied to the bath, and recirculated (total volume: 12.5 ml; ow rate: 6–8 ml/min). Following bath-in of Rubi-Glu (< 2 min), the holding potential was increased to 40 mV. NMDA currents were measured from 20 to 50 ms post-pho- tolysis stimuli. As the amplitude of the current produced in response to hyperpolarizing membrane voltage steps (< 5 mV) was small, the mEPSCs were then depolarised to −30 mV, and then depolarised to −30 mV, in order to destripe the summation properties of dendrites in L4 SCs we performed near- simultaneous dendritic spine 2-photon glutamate uncaging. Near-simultaneous dendritic spine 2-photon glutamate uncaging was achieved by placing a field electrode (a patch electrode filled with ACSF in visually identified barrels and stimulating the thalamus. When a field response was observed, the cell was switched to current-clamp mode and set to optimal mEPSCs at each holding potential using the event-detection filter of the ampli- tude. Data were shown as the average ± SEM of three experiments. Near-simultaneous dendritic spine 2-photon glutamate uncaging was achieved by placing a field electrode (a patch electrode filled with ACSF in visually identified barrels and stimulating the thalamus. When a field response was observed, the cell was switched to current-clamp mode and set to optimal mEPSCs at each holding potential using the event-detection filter of the ampli- tude. Data were shown as the average ± SEM of three experiments.
and slices embedded within OCT prior to sectioning. The OCT block containing the recorded slice was trimmed to the slice surface and then 50 µm sections taken from the block. Sections were not dehydrated or post-fixed and then incubated with streptavidin conjugated to AlexaFluor 488 (1:500, Invitrogen, UK) at 4 °C for 3–5 days. The slices were then washed for 2 h in repeated washes of PBS, and then desalted with PB and mounted on glass slides with fluorescence pro-
tecting mounting medium (Vectorshield, Vector Labs, UK).

Sections were imaged on a gated-stimulated emission-depletion (STED) microscope (SP8 gSTED, Leica, Germany). Cells were found using epifluorescent illumination (488 nm excitation) under direct optics at low magnification (∗×20 air immersion objective lens, N.A. 0.75) and then positioned under high magnification (∗×100 oil immersion objective lens, N.A. 1.4, Olympus, Japan) and then switched to gSTED imaging. Sections were illuminated with 488 nm light, produced by a continuous-wave laser, and short sections of non-uncaged dendrite used to optimize acquisition parameters, first under conventional confocal detection, then by gSTED imaging. The 488 nm illumination laser was set to 60–70% of maximum power, and the continuous-wave STED laser (592 nm) set to 25% and gated according to the best STED-depletion achievable in the samples (1.5–8 ms gating). Once optimized, a region of interest (ROI) was selected over the uncaged dendrite, which at 1024 × 1024 pixel size, gave a pixel resolution of 20–30 nm. Short stacks were taken over dendritic sections containing uncaged and non-spines (0.5 µm steps) while STED images interleaved with 10 times faster frames for comparison of STED effect. STED images were deconvolved (Huygens’ STED option, Scientific Volume Imaging, Netherlands) and uncaged spines identified by comparison to live 2-photon images (see Fig. 2a). Measurements of head width and neck length were then made on the deconvolved images in FIJI (ImageJ)36.

Serial block-face scanning-electron microscopy (SBF-SEM) of L4 SCs. For SBF-SEM, 10 P14 mice (3 WT/7 Fmr1−/−) were perfused after sacrifice. Briefly, mice were sedated with isoflurane and terminally anaesthetized with IP sodium pentobarbital (50 mg/mouse). The chest was opened and 10 ml of PBS (pH 7.4, filtered) trans-
cardially perfused (∗0.5 ml/second); once cleared the PBS was replaced with ice-cold fixative solution containing (3.5% PFA, 0.5% glutaraldehyde, and 15% saturated picric acid; pH 7.4), and 20 ml perfused. Brains were then removed and post-fixed overnight at 4 °C in the same fixative solution. 60 µm thick coronal sections were cut on a vibratome (Leica VT1000) and S1 identi-

Reporting summary. Further information on research design is available in the Nature Research Reporting Summary linked to this article.

Data availability
All datasets will be made available upon reasonable request.

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Author contributions

S.A.: designed and performed experiments, analysed/interpreted data and wrote the paper; A.P.F.D.: designed and interpreted, performed experiments, analysed data and wrote the paper; O.R.D.: analysed/interpreted data and wrote the paper; A.J.: performed experiments, analysed/interpreted data; J.T.R.L.: designed experiments and wrote the paper; G.E.H.: analysed/interpreted data, obtained funding and wrote the paper; D.J.A.W.: designed experiments, analysed/interpreted data, obtained funding and wrote the paper; P.C.K.: designed experiments, analysed/interpreted data, obtained funding and wrote the paper.

Additional information

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Competing interests

The authors declare no competing interests.

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